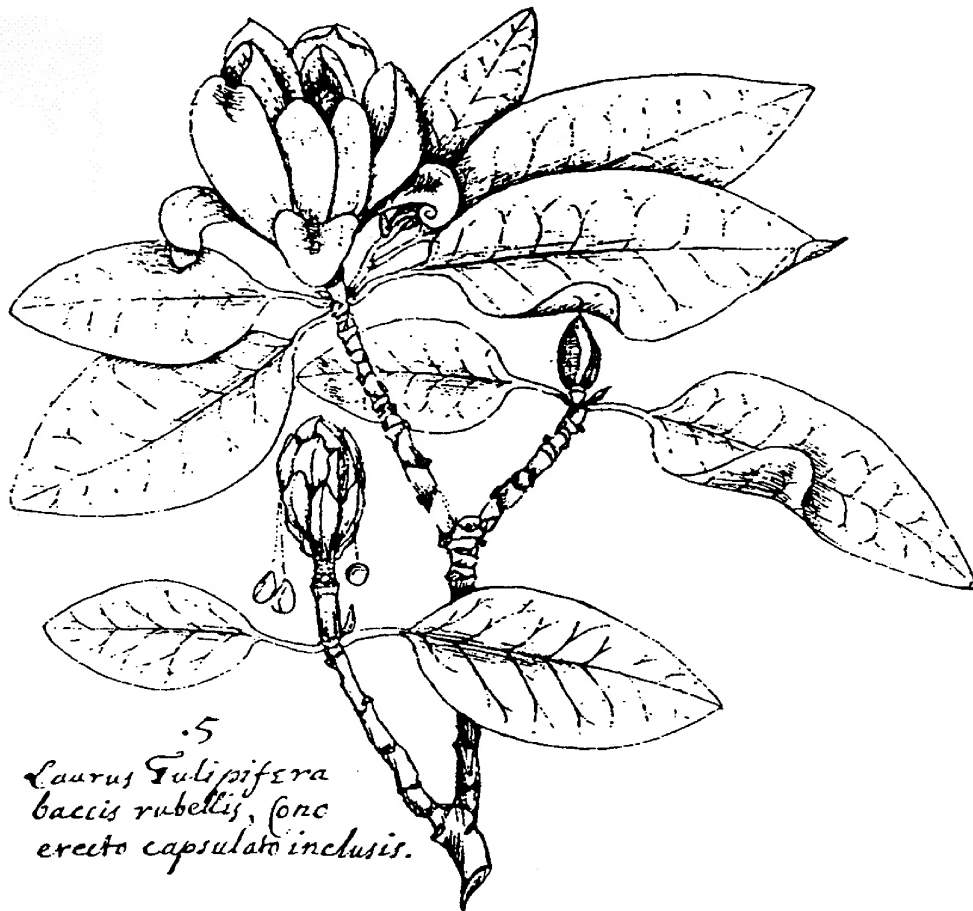


BANISTERIA

A JOURNAL DEVOTED TO THE NATURAL HISTORY OF VIRGINIA



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Back cover: Anemone quinquefolia Linnaeus (Wood Anemone). Original drawing by John Banister; sent to Bishop D. H. Compton in 1689. Figure 72 in folio in Sir Hans Sloane's MS 4002 in the British Museum.

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Natural History of the Eastern Harvest Mouse in Southeastern Virginia

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ABSTRACT

The Eastern Harvest Mouse, *Reithrodontomys humulis*, has been studied extensively in southeastern Virginia since 1979, using a combination of live and pitfall trapping methods. This smallest rodent of eastern North America also is one of most versatile, occupying a range of habitats in southeastern Virginia from old fields in different stages of succession, brushy edges, and forests of different types. As with other species of *Reithrodontomys*, *R. humulis* often is associated with the Hispid Cotton Rat, *Sigmodon hispidus*, with both reaching modest densities in old fields. Two capture-mark-release studies of small mammal communities in southern Chesapeake lasting eight and nine years revealed that the Eastern Harvest Mouse was third in total abundance, behind Hispid Cotton Rat and Meadow Vole, *Microtus pennsylvanicus*, as old fields transitioned into forests. Multiple field studies using pitfall traps in a range of habitats in southeastern Virginia also indicated that harvest mice often arrive early in succession and stay later than other rodents.

Keywords: Coastal Plain, Eastern Harvest Mouse, habitat selection, *Reithrodontomys humulis*, small mammals.

INTRODUCTION

The Eastern Harvest Mouse, *Reithrodontomys humulis*, is a cricetid rodent with a distribution mostly in the southeastern U.S. (Stalling, 1997). With adults averaging about 8 g, this is the smallest rodent in the eastern U.S. Its small size alone distinguishes it from the 16–25 g White-Footed Mouse, *Peromyscus leucopus*, the native rodent with which adults are most comparable in coloration and body form; both have brownish backs, white or nearly white underbellies, and long tails. The other similar small mouse with which *R. humulis* might be compared is the House Mouse, *Mus musculus*, which has large and naked ears, a nearly hairless unicolored tail, and a gray or orangish underbelly. Their behaviors differ too; when placed in a bucket after removal from a live trap, an Eastern Harvest Mouse is likely to remain calm and groom itself or eat seeds, whereas a House Mouse is frenetic, running and leaping in its attempts to escape.

Much of the information in this report comes from the work of Old Dominion University graduate students conducting field research projects as part of their thesis research for the Master of Science degree. Jean Ferguson Stankavich, who conducted capture-mark-release (CMR) studies of small mammals in the northwest section of the Great Dismal Swamp National Wildlife Refuge, found Eastern Harvest Mice to be numerically dominant in two CMR grids. Sarah Crawford added an analysis of vegetation composition and structure to her study of small mammal communities with harvest mice. Michelle Cawthorn Chandler used a specially built trap, with a 2.1 by 2.1 cm opening, to exclude larger small mammals in an effort to study the smallest members of the small mammal community in an old field. These studies were conducted in habitats in early stages of succession, where densities of small mammals tend to be highest. Additional information about distribution and relative abundance comes from field studies using pitfall traps on dozens of 0.25 ha grids that enable comparisons

of relative densities among habitat types and from two long-term CMR studies of rodent communities.

GENERAL CHARACTERISTICS

The Eastern Harvest Mouse is grayish brown with a darker mid-dorsal stripe on the back, with lighter and sometimes rusty sides, and whitish feet (Fig. 1). The tail is about the same length as the head-body length; in a series of harvest mice from Isle of Wight County measured by the author, the tail was 47.8% of total length for 32 males and 47.6% of total length for 30 females. The underside of the tail is whitish, as is the belly. The eye is large and dark, suggesting nocturnal behavior. The vibrissae (whiskers) are numerous, long, and pale at the tips. Another feature that distinguishes the Eastern Harvest Mouse from other long-tailed rodents in southeastern Virginia is the groove in the upper incisor, which can be seen with the naked eye. The anterior face of the incisor is folded, giving it a corrugated appearance. The function of this feature is unknown, but the fold probably strengthens the tooth, thus reducing the likelihood that the tooth will break when opening hard-coated seeds. The sexes are similar in size (Dunaway, 1968), but when weighed with a 10-g Pesola scale (with 0.2 g calibrations) non-pregnant females from Chesapeake, Virginia ($\bar{x} = 8.20 \pm 0.3$ SE g, $n = 35$) weighed significantly ($p < 0.05$) more than male harvest mice ($\bar{x} = 7.04 \pm 0.1$ SE g, $n = 42$) (Cawthorn & Rose, 1989). The weight of males was relatively constant throughout the year but weights of females peaked in autumn, suggesting that as the season of greatest reproduction.



Fig. 1. An adult eastern harvest mouse, *Reithrodontomys humulis*. Photo credit to West Virginia University Wildlife and Fisheries Science study guide (Edwards).

DISTRIBUTION

The Eastern Harvest Mouse has a mostly southeastern distribution in the US, extending eastward from eastern Oklahoma and Texas to states lying south of the Ohio River, but also including southern Ohio, western Maryland, all of Virginia, and points southward. It may be absent from the southern tip of Florida. Some populations in Texas, Oklahoma, Arkansas, and Louisiana overlap in distribution with those of *R. fulvescens*, a larger species that has been studied extensively in the Texas coastal prairies by Cameron (1977).

Information on the distribution of *R. humulis* is accumulating as more community studies are being published, so the map of Stalling (1997), already an improvement of Hall (1981), continues to be revised. For example, before 1988, *R. humulis* was known from only three counties in Oklahoma and was considered a rare mammal, but by 2011, its presence had been recorded in six more counties (Braun et al., 2011).

Three subspecies are recognized. Howell (1940) described *R. humulis virginianus* based on specimens from Amelia County, located in central Virginia just one county southwest of Richmond. This subspecies, present in the eastern half of Virginia, is paler and more grayish, with a blackish-brown mid-dorsal stripe, and with white feet compared to *R. h. humulis*, the other subspecies east of the Mississippi River. *R. h. merriami* is present in the four western states. As presently understood, the northern distribution of coastal populations of *R. humulis* is in southeastern Virginia. Field studies of the Eastern Shore of Virginia by Rose and colleagues (e.g., Rose & March, 2013) have failed to record any *R. humulis* in either Northampton or Accomack counties, so its movement northward likely is blocked by the Chesapeake Bay. Pagels & Moncrief (2015) also consider *R. humulis* to be absent on the Eastern Shore.

FORM AND FUNCTION

Its small size and long tail suggest that this rodent can climb into even herbaceous vegetation, perhaps to glean seeds or capture insects. Relatively little is known about its diet, except that it eats some seeds. But *R. humulis* is not considered to be truly arboreal because its softball-sized grassy nests are placed in low herbaceous vegetation or on the ground rather than in tree holes, as truly arboreal rodents usually do.

The monthly mean weights of males from southeastern Virginia were relatively stable (Chandler, 1984). In Tennessee, unbred lab-reared adult harvest

mice of both sexes had identical weights, 8.2 g (Dunaway, 1968) and Kaye (1961) reported that lab-reared 50-day-old adult males and females weighed the same. By contrast, field-caught females were heavier than males in all but 3 of 21 months in Tennessee (Dunaway, 1968), suggesting that pregnancy accounts for most differences in weights of the sexes. As in southeastern Virginia, the weights of males were fairly constant throughout the year (Dunaway, 1968). In brief, adults are similar in size, about 8 g, and of equal body length.

Small body size means that, on a per gram basis, *R. humulis* has a higher metabolic rate, and thus relatively higher energy requirements, than larger mammals, a relationship recognized decades ago by Kleiber (1961). Furthermore, below the temperature zone of least energy cost, termed thermoneutrality, the energetic costs increase disproportionately. For example, the resting metabolism of the Eastern Harvest Mouse at 23° C is 4.35 ml of oxygen per gram of body weight per hour, but at 7° C the metabolic rate more than doubles, to 9.62 ml of oxygen per gram per hour; the comparable values for the twice-as-large White-footed Mouse are 3.04 and 5.68 (Dunaway, 1968). Also, because of its small size, it can neither reduce heat loss via long and dense insulative fur nor accumulate large fat reserves, two ways larger mammals can conserve or produce heat during periods of cold temperatures. These factors likely restrict the distribution of *Reithrodontomys*, a genus with tropical origins, to sub-tropical and temperate climate zones in the US.

The numbers of red blood cells per unit volume were similar to those of larger rodents examined by Dunaway (1968). However, harvest mice had much higher concentrations of hemoglobin (g/ml) in the erythrocytes than in larger rodents, likely an adaptation to deliver sufficient oxygen to cells of a small mammal with high metabolic rate.

REPRODUCTION

The breeding season for *R. humulis* likely varies by geographical location, starting earlier in spring in southern than in northern populations. In southeastern Virginia, breeding peaks were observed in spring and autumn, with a lull in summer (Cawthorn & Rose, 1989). The higher body weights of females plus the many gray-backed juveniles indicate greater levels of reproduction in autumn than in spring.

Studies in the laboratory indicate that females in late pregnancy become intolerant of males and that males take no role in parenting (Kaye, 1961), the pattern seen in most mammals. Near the end of the 21-day gestation period, the female builds a birthing nest of dried grasses,

in which the young are reared for about three weeks. Litter size averaged 2.2 for nine lab females in Florida (Layne, 1959) but was 3.2 for nine lab females from North Carolina (Kaye, 1961). Later, Dunaway (1962) reported finding three litters of three and three litters of four born in live traps in Tennessee; he also took a 17-g female into the lab where two days later it gave birth to eight young, the weights of which totaled nearly 8 g. Taken together, the litter size is about three; these are weaned near the end of the third week of life, at weights of about 5 g, the lightest animals trapped in most studies. On 16 December 2018, I recorded a 12-g pregnant female with partially open pubic symphysis and enlarged nipples, indicating that this female produced a late litter in southeastern Virginia. In my experience, females heavier than 10 g are pregnant.

Few details are known about reproduction in male eastern harvest mice, in part because indicators of reproduction are fewer than in females. During the breeding season the enlarged testes are descended into the scrotum, and such males are judged to be reproductive. In the winter non-breeding season, the testes of many mammals, including harvest mice, decrease dramatically in size, often losing 95% of their weight, and such males are non-reproductive. Cawthorn & Rose (1989) observed scrotal males in every month of the year, with lowest rates (10%) in winter. In the nearby Great Dismal Swamp, Stankavich (1984) also found some scrotal males in winter (24%), suggesting the possibility of occasional year-round breeding in southeastern Virginia. Coastal Virginia averages 10 cm of snow, 10–20 nights below 0° C, and short periods of frozen soil. By contrast, no scrotal males were observed in December, January, and February in Tennessee (Dunaway, 1968). These observations suggest that populations in southern states (or coastal locations in Virginia) might have year-round reproduction, although it was not observed by Layne (1959) in Florida. In South Carolina, highest numbers of captures were recorded in January, indicating that breeding levels were greatest in late autumn (Briese & Smith, 1974).

ECOLOGY

Much new information about *R. humulis* in southeastern Virginia has been published in recent decades by using CMR methods on small square or rectangular (row by column) grids with live traps placed at the coordinates. At monthly or twice-monthly intervals, the traps are baited and 'run' for three consecutive days. Each captured animal is given a unique number, usually with an ear tag, weighed, and its sex and reproductive condition are recorded. The animal is then released at the point of capture. The goal is to trap

such tagged animals in successive months and record the events of their lives: features such as their changes in body mass, levels of reproduction, rates of body growth and survival, area of use on the grid, among others. Also, the vegetation of the grid often is studied, both for its list of plant species but also for details of plant contacts at different heights in an effort to learn whether the vertical structure of the plant community is more or less important than the presence of certain plant species. For example, when grasses dominate the plant community, vertical structure is dense with stems and leaves below about 0.5 m. Later in biological succession, when shrubs and trees are common, the density of vegetation near the ground surface is much less, but vertical elements are more common, increasing vegetation complexity in a different way.

The first CMR study in southeastern Virginia was conducted under a 40-m wide powerline in the northwest section of the Great Dismal Swamp National Wildlife Refuge (Stankavich, 1984). Two study grids of Fitch live traps (Rose, 1994) were established in habitat dominated by plants typical of early successional stages in a swamp: grasses and forbs, and in wetter places, sedges, rushes, and spikerushes. Some deciduous trees and shrubs were present too, especially in the slightly higher places where winter flooding did not kill them. Harvest mice were the most common small mammal in this habitat, comprising 71 of 155 total individuals (Rose & Stankavich, 2008). In an 18-month CMR study, conducted in Suffolk just west of the Great Dismal Swamp National Wildlife Refuge, seven harvest mice were tagged, along with 47 *Synaptomys cooperi* (Southern Bog Lemming) and 110 *Microtus (Pitymys) pinetorum*; no other rodents were captured in this community where minor species dominated (Rose & Ford, 2012).

Michelle Cawthorn (Chandler, 1984) conducted CMR trapping of small mammals every other week for a year on two grids in an old field in the Bowers Hill region of Chesapeake. The tiny specially built traps excluded adults of the common small mammals and thus she caught mostly Eastern Harvest Mice and House Mice, 51% and 39%, respectively, of 703 total captures (Cawthorn & Rose, 1989). Highest densities for *R. humulis* were achieved on both grids in autumn and winter, with 44 harvest mice per hectare; the average density was 21.9 and 21.8/ha on both grids across the study. The adult mortality rate of 6 percent per month was constant for the year-long study. Home range, the area of greatest use, was similar for both sexes, at about 1000 m². Lifespans, based on three or more captures, averaged about 10 weeks for both sexes, which if added to the 20–30 days for newborns to become trappable, equates to mean lifespans of about 100 days (Cawthorn & Rose, 1989), similar to those (90–120 days) in

Tennessee (Dunaway, 1968).

Cawthorn/Chandler, (1984) recorded 29 plant species on Grid 1 and 27 species on Grid 2, 18 of which were present on both grids; asters dominated on Grid 1 but honeysuckles (*Lonicera*) on Grid 2. But height of vegetation was more important than species composition, a conclusion also reached by Crawford (2013), who used assessments of plant composition and measurements of structure while trying to understand the strong association of harvest mice with the Hispid Cotton Rat (*Sigmodon hispidus*), adults of which are mostly 80–120 g in southeastern Virginia. Numerous studies report that *S. hispidus* and the local *Reithrodontomys* species often occur together, regardless of the region. Both are tropical genera with populations in the US at the northern limits of distribution (e.g., Braun et al. [2011]; Brady & Slade [2001] for *R. megalotis* in eastern Kansas; Rose et al. [2018] for *R. humulis* in southeastern Virginia). Both genera reach highest densities in old field and other early successional habitats, but the reasons for their frequent coexistence remain unclear, whether by being active at different times of day, by mutual avoidance at the microhabitat level, or by differential use of resources (Crawford, 2013).

Using live-trapping records, Crawford (2013) found no evidence that either harvest mice or cotton rats avoided the other on either of two 1-ha grids, each trapped monthly for multiple years. A negative association between captures at each station was recorded for only one month over that period. Both species tended to occupy areas with few or no trees, and harvest mice were more likely than cotton rats to be present if the open sites were wet. Both species tended to use areas with dense vegetation near the ground surface, regardless of plant species composition. Crawford speculated that differential use of resources (harvest mice are primarily seed-eaters whereas cotton rats eat stems and leaves, mostly of monocots) and the broader habitat tolerances of harvest mice as the most likely reasons for the coexistence of these two species in southeastern Virginia.

In evaluating the changes in composition of the community of small mammals on the same two grids analyzed by Crawford (2013), Rose et al. (2018) found that harvest mice and cotton rats were early colonizers in grassy old fields in the third year after a farm field was abandoned and both species persisted while other community members came and went. Across eight years of study on one site and nine years on the other, *R. humulis* was third in total abundance on both grids, with cotton rats being most numerous on one grid and Meadow Voles (*Microtus pennsylvanicus*) numerically dominant on the other. Thus, although many investigators would consider harvest mice to be a minor

species in the community of small mammals, in southeastern Virginia they are early arrivals, third in abundance during succession, and they are still present at the point when the forest small mammals, such as White-footed Mice and Golden Mice (*Ochrotomys nuttalli*), arrive and become the dominant rodents.

In field studies using pitfall traps on 0.25 ha grids, the results were similar. For example, *R. humulis* was present on 13 of 14 grids in Isle of Wight County, more than any other species, and was second in abundance to Least Shrew (*Cryptotis parva*) (Rose, 2005). Similar results were found in 19 pitfall grids in Virginia Beach, Chesapeake, and Suffolk (Rose, 2016).

In his pitfall-trapping study of small mammals in and near the Great Dismal Swamp National Wildlife Refuge, Everton (1985) found *R. humulis* on 10 of 21 one-quarter hectare grids, and fourth in overall abundance behind two shrews (Southeastern Shrew, *Sorex longirostris*, and Least Shrew) and Southern Bog Lemmings (*Synaptomys cooperi*). In the analysis of vegetation structure, Everton found that *R. humulis* was associated with high values for stem densities from ground level to 40 cm and for average height of herbaceous vegetation, indicating a strong preference for dense cover of plants, mostly grasses, near the surface. In a summary of studies of small mammals conducted across the range of habitats in the Great Dismal Swamp, using all trapping methods (live, pitfall, and break-back traps [used in the late 19th century]), *R. humulis* was third in total abundance, behind Short-tailed Shrews (*Blarina* spp.) and White-Footed Mouse (Rose et al., 1990, Table 4).

Thus, in southeastern Virginia at least, *R. humulis* is the most versatile rodent in the small mammal community. For example, one was caught on a tall sand dune at Little Creek Amphibious Base in Norfolk, along with House Mice and White-footed Mice (Rose & Sweitzer, 2013).

More commonly, *R. humulis* arrives early in old fields dominated by grasses and forbs, sometimes sharing early arrival status with house mice. Soon other species, such as herbivores as cotton rats, meadow voles, and rice rats, arrive and some of these become dominant species for months or years. But when the herbaceous vegetation thins and eventually is shaded out by shrubs, saplings, and trees, the herbivorous rodents disappear, often quickly. Based on studies of two old fields going through succession, cotton rats and harvest mice often were still present before forest rodents come to dominance. Eastern Harvest Mice are much less common in the forests of southeastern Virginia than in earlier stages of succession, but often they are present in small numbers (e.g., Everton, 1985). Others also have found *R. humulis* in forests, such as in wetland forests in Tombigbee

National Forest in Mississippi (Edwards & Jones, 2014), and rarely in pine forests (Dolan & Rose 2007). In their pitfall trapping study in the upper coastal plain of Virginia, Bellows et al. (2001) found *R. humulis* to be more abundant in old field habitats than in other macrohabitats; harvest mice were present in oak-hickory forest and young pine forests, but not in older pine or oak-pine forests. In a four-year study in the North Carolina coastal plain, *R. humulis* had good recruitment and survival in all five treatments that provided varying amounts of structure (woody debris, pine seedlings, switchgrass), and by year four it outnumbered the other three colonizing species (Homyack et al., 2014). In brief, numerous studies reveal *R. humulis* to be versatile by occupying a range of habitat types.

BEHAVIOR

Harvest mice are primarily nocturnal, and thus are active during the coldest part of the day, enabling them to benefit from the heat generated during foraging and other activities. At thermoneutrality (22° C), *R. humulis* shows an innate increase in metabolic rate at the approach of darkness, as if foretelling the beginning of nocturnal behavior (Baker, 1974). Baker, who measured CO₂ production rather than oxygen consumption, also recorded a doubling of metabolic rate when harvest mice were housed at 9° C.

Nocturnal behavior means that owls are their main avian predators, as recorded by Klippel & Parmalee (1982) in their study of pellets from a wintering Long-eared Owl (*Asio otus*) in the Nashville Basin of Tennessee. *R. humulis* was second in abundance ($n = 78$) to Prairie Vole (*Microtus ochrogaster*, $n = 129$) among 71 complete pellets. In a study conducted near Williamsburg, Virginia, Rosenburg (1986), who followed tagged Barn Owls (*Tyto alba*) via radio-tracking, found small numbers of Eastern Harvest Mice in their pellets in most seasons. The Meadow Vole, also common in old fields in early succession, was the main food of these owls.

The observation of multiple captures in live traps suggests some degree of sociality in harvest mice. In southeastern Virginia, 6.4% of total captures were as multiple captures, with more male-male pairs and fewer female-female pairs than expected (Cawthorn & Rose, 1989). Others have reported huddling, especially during winter months. For example, Dunaway (1968) reported that only 3 of 18 eastern harvest mice were alone in the nest cans of traps on a late January day; the others were in groups of 6, 4, 3, and 2. I have observed similar social groupings in *R. megalotis* in eastern Kansas, instances in which up to 11 adults shared grassy nests in gallon-sized nest chambers. Similar social groupings were observed

in *R. fulvescens* in the Texas coastal plain (Spencer et al., 1982). Formation of social groups is especially important for tiny mammals, enabling them to share the costs of staying warm together in their well-insulated grass nests. In his analysis of spacing behavior among individuals of *R. humulis*, Dunaway (1968) found little evidence of territoriality: territorial individuals are anti-social.

One consequence of social groupings is the potential for the ‘sharing’ of ectoparasites. Clark & Durden (2002) found 10% prevalence both of fleas (*Polygenis gwyni*) and of ticks (*Amblyoma maculatum*) in Eastern Harvest Mice in Mississippi. In southeastern Virginia, of nine small mammal species evaluated for ticks over a period of years, harvest mice had the lowest proportion of infestation; 18.3% had ticks, mostly on the ears (17 of 93; H. Gaff, pers. comm.). By contrast, another benefit of social groupings is allogrooming, i.e., the removal of ectoparasites by other members of the group. There is no direct evidence of allogrooming in *R. humulis*, but the low percentage of ticks on harvest mice is consistent with this hypothesis.

The killing of young by siblings or mother seems to be a common behavior, at least in captivity; sometimes this unexplained behavior is followed by cannibalism (Dunaway, 1962; Kaye, 1961).

GENETICS

Information on the chromosomes of *R.* and its congeners is mostly derived from studies conducted nearly 30 years ago. Carleton & Myers (1979) reported that *R. humulis* had a diploid number of $2n = 51$ for two females (no males were assessed); the chromosomes were mostly small-to-medium acrocentrics plus five pairs of larger and bi-armed chromosomes. The unpaired element was a small metacentric chromosome. The $2n = 51$ was confirmed by Robbins & Baker (1980), and although they determined the FN to be 78, they could not determine the origin of the unpaired element. Much remains to be learned about the genetics of *R. humulis* and others in this genus.

CONSERVATION STATUS

The 2016 International Union for the Conservation of Nature Red List of Threatened Species lists *R. humulis* as “Least Concern,” and with stable populations. However, a map on the same website indicates that Oklahoma considers the species to be “critically imperiled,” but this statement conflicts with Braun et al. (2011), which adds six counties to their known locations in the state. The map also lists the species as “possibly extirpated” in West Virginia and “not ranked” or “under

review in Mississippi, South Carolina, and Florida. The Ohio Department of Natural Resources website states that *R. humulis* is a “Threatened” species in their state, despite Harder et al. (2014). The other states, including Virginia, assess their populations to be “secure” or “apparently secure.”

REMARKS

The name *Reithrodontomys humilis* was given in 1841 by John James Audubon and his son-in-law, John Bachman, based on specimens collected near Charleston, South Carolina. Early in the next decade, these same authors, far better remembered for their studies of and naming of many North American birds, published *Quadrupeds of North America*, the first comprehensive book on New World mammals. They chose the genus name, *Reithrodontomys*, derived from three Greek words (Lowery, 1974), because of the grooved incisor: *reithron* (groove), *odous* (tooth), and *mys* (mouse). The specific name *humilis* may be a misspelling of *humilis*, which means “little harvest mouse.” In their *Quadrupeds* book, the authors used the latter spelling. The tiny mouse of western Europe and the British Isles is also called “harvest mouse,” but it is in a different genus, *Micromys*, literally “tiny mouse.”

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Investigating Campus Features that Influence Bird-window Collisions at Radford University, Virginia

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ABSTRACT

Window collisions pose a serious risk to birds, second only to domestic/feral cats. We sought to quantify the impacts of this threat at Radford University, a campus situated within a rural landscape and along a major migratory route (New River). We searched for evidence of bird-window collisions (BWCs) at 15 buildings in 2018 and 2019. In nearly 1,000 hours of surveys we discovered 51 birds (23 species) thought to result from BWCs. Increased window area tracked with a greater number of mortalities/building. Building height and surrounding vegetation metrics were not significantly related to BWCs. Species' residency status did not significantly influence mortality events. Compared to BWC surveys nationwide, our number of mortalities was low, especially relative to our substantial surveying effort. Although this finding might suggest that Radford University buildings are not a significant source of mortality for birds, we recognize that (1) *a priori* surveying biases likely underestimated actual mortalities, and that (2) Radford University's architectural changes in the last several years are increasing the likelihood of BWCs in the future. We suggest that Radford University explore window decals on current windows and incorporate "bird-friendly" glass on aspects that comprise large proportions of glass. Both of these steps contribute to Radford University's goal of increasing the number of LEED-certified buildings on campus.

Keywords: avian migration, avian mortality, building height, deterrents, time-of-day, vegetation, window area.

INTRODUCTION

Bird-window collisions (BWCs) are a substantial anthropogenic source of bird mortality, accounting for an estimated 365–988 million bird deaths annually. BWCs are the second largest cause of bird deaths, behind domestic and feral cats (*Felis catus*; Loss et al., 2014; Kahle et al., 2016). Collisions typically are not limited to a particular avian taxon, and they can negatively affect common birds as well as species of conservation concern (Loss et al., 2014; Hager et al., 2017).

Previous BWC studies have covered the gamut of building scenarios, from high-rises in a metropolis (Chicago: Briscoe & Dampier, 2019; Manhattan: Gelb & Delacretaz, 2009) to myriad college campuses (Hager et al., 2017). Multiple studies have investigated landscape and geographic metrics, as well as species-specific natural history features that may significantly affect the

likelihood of BWCs. Features of the buildings, such as total window area and building height have been analyzed in several studies. While Bayne et al. (2012) found that collision rates were higher in rural areas where building density was lowest (Alberta, Canada), multiple studies found the opposite trend – higher building densities resulting in higher collision rates (Loss et al., 2014; Schneider et al., 2018). Hager et al. (2017) explained this difference along the spectrum of land development: building height and window area had a proportionally larger influence on BWCs in rural areas than in urban areas. This difference was most apparent during peak migration times, as non-resident birds were more likely selecting rural landscapes as suitable routes, and low-density buildings had a proportionally greater number of BWCs than buildings of the same size (height, window area) in an urban landscape. Artificial light also may increase BWCs, as nocturnal migrants may be

confused by and attracted to them in flight (Hager et al., 2017).

Features immediately surrounding buildings also have been studied as potential predictors of window collisions. Because surfaces like windows may reflect images of nearby vegetation, birds are likely confused, seeing it instead as a perching site or other suitable habitat (Hager et al., 2017). In many studies, vegetation effects were significant, but never primary metrics that influenced BWCs. Qualitative measures of vegetation have been implemented, but categories varied by study. The presence/absence (Chin, 2016) of vegetation was one method, while others loosely categorized density, using “some” vs. “extensive” vegetation (Gelb & Delacretaz, 2009) or “vegetated” vs. “less vegetated” (Gelb & Delacretaz, 2006). Quantitative vegetation metrics have covered the gamut of methods, but many used broad-scale vegetation metrics that relied on existing GIS-based layers. For example, Hager et al. (2017) utilized percent “vegetation” within 50 m of a building, while Schneider et al. (2018) examined percent landcover class within the same radius (e.g., lawn, trees, ornamental vegetation). Quantitative vegetation measures *in situ* were utilized less often, and were not always collected by the authors (e.g., Kummer et al. [2016a] asked citizen scientists to report the average height of vegetation around their residence). It is clear that no consistent method to measure vegetation exists for BWC studies. In these referenced studies, vegetation seems to have no more than a secondary effect on BWCs (typically less influential than window area or generalized building structure). Although avian and mammalian studies not focused on BWCs have successfully utilized metrics such as total vegetation volume and the Levins diversity index to quantify vegetation *in situ* (Francel & Schnell, 2002; Leighton et al., 2009), to our knowledge, no BWC studies have utilized these quantitative on-the-ground metrics to encapsulate vegetation effects. However, there is evidence that metrics are related to bird community composition (Mills et al., 1991; Francel & Schnell, 2002).

Aspects of avian natural history may compound the anthropogenic/structural effects described above. In temperate regions, timing of migration (if the species migrates) and circadian activity patterns may affect the chance of BWCs. Kahle et al. (2016) found that BWCs increased during the periods of migration (April and October) and in mid-summer (July) when most birds are breeding. Numerous studies also concluded that BWCs were greatest during migration periods (Johnson & Hudson, 1976; Codoner, 1995; O’Connell, 2001; Gleb & Delacretaz, 2006; Hager et al., 2008). Despite the seasonal timing of these collisions, Klem (1989) concluded migratory status (as well as sex, age, and

weather) did not affect BWCs. Supporting this, Blem & Willis (1998) and Kahle et al. (2016) concluded that migrating birds may not be major contributors to collisions. An examination of circadian activity patterns also presented us with conflicting data. While time of day was not a significant factor for BWCs by Klem (1989), others found clear trends in timing of collisions across daylight hours. Kahle et al. (2016) studied BWCs in an urban park (Golden Gate Park, San Francisco, California) and found that the greatest number of strikes occurred during mid-morning hours, 0900 – 1100 h. They found a steady decline in collisions throughout daylight hours. However, 83% of their documented BWCs occurred in daylight hours, as their pre-0900 h (early morning) surveys documented just 17% of the collisions. Other BWC studies concluded *a priori* that collisions were more likely during daylight hours, and limited their carcass surveys to afternoon time periods (e.g., 1400 – 1600 h; Hager & Consentino, 2014; Hager et al., 2017).

In Virginia, BWCs have been investigated at a coastal campus at Old Dominion University (as part of a 40-campus national survey, individual results not presented in Hager et al., 2017) and in the western montane regions at the Virginia Tech Corporate Research Center (VTCRC) in Blacksburg. Although not the university campus proper, VTCRC does include 28 buildings (mostly 2-story, with maximum window areas of 693 m²) across 93 ha. In their study, they documented 240 bird casualties across 298 survey days. They discovered more BWCs with increased window area and an increase in ornamental vegetation around the buildings (Schneider et al., 2018). From this publication stemmed questions about nearby Radford University – a suburban campus set in a rural landscape – similar in land area to VTCRC but with taller buildings at greater densities.

Radford University, an 82.6-ha campus (37.13870°N, 80.55759°W; Fig. 1), is situated along a recognized migratory bird highway, the New River (e.g., VDGIF includes portions of the New River on its Bird & Wildlife Trail networks, VDGIF, 2019). Located about 20 km southwest of VTCRC, this campus includes >30 buildings that are 3-4 stories tall, and one residence hall that is 13 stories tall. Construction on new and renovated buildings occurs year-round. Although the university seeks to build or renovate buildings so that they are LEED-certified, no buildings to date have incorporated bird-detering windows or bird-detering window decals (M. Biscotte, Office of Planning and Construction, Radford University). Furthermore, windows have been a substantial (window area > 1500 m²) component of facades at new or renovated buildings along major thoroughfares (Center for the Sciences, College of

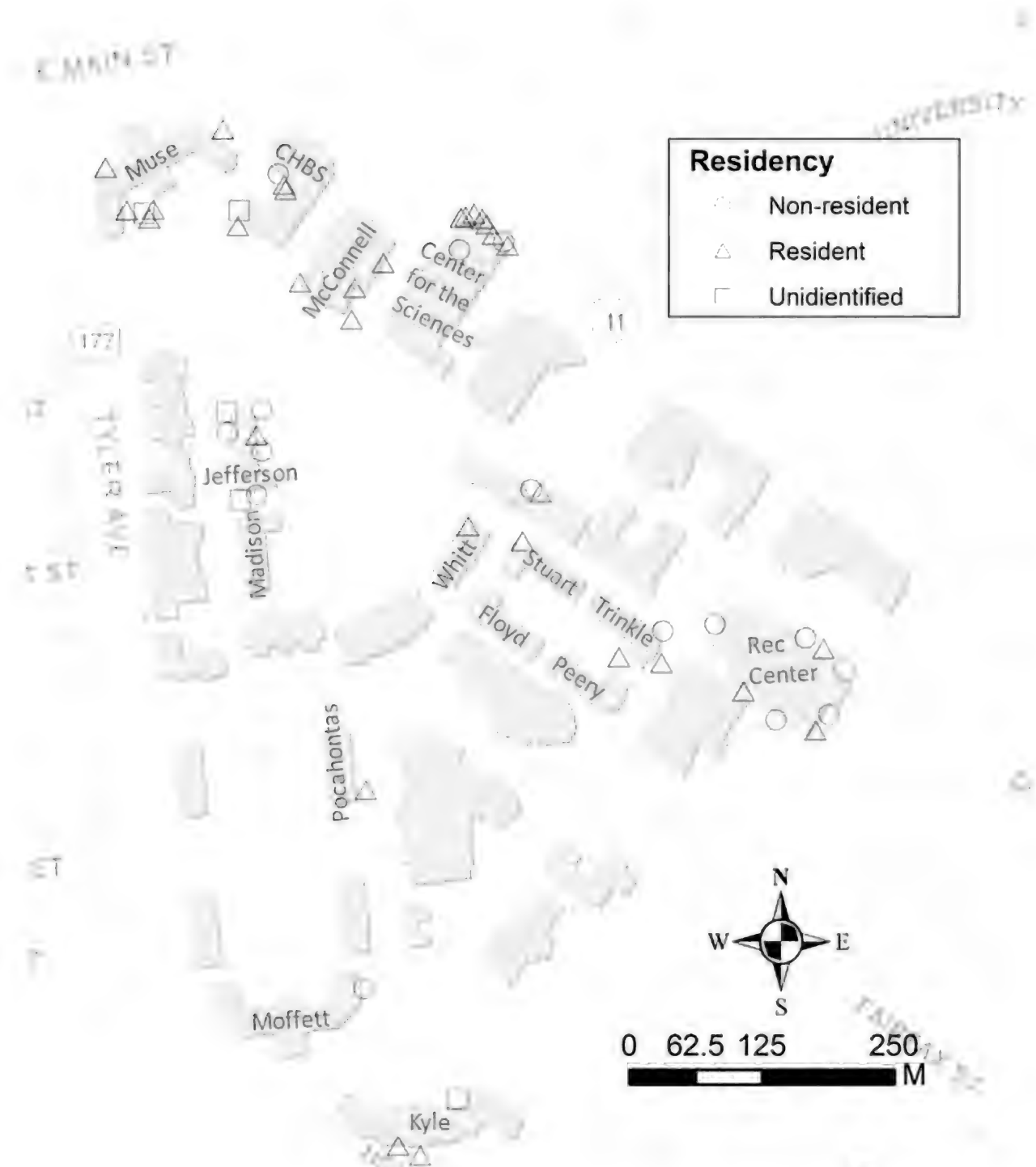


Fig. 1. Radford University, Virginia campus map, with 15 buildings (labeled with names, in red) surveyed for bird-window collisions in 2018 and 2019. We recorded 0 – 8 hits/building. Locations of 51 documented BWC casualties, identified by species classified by residency status: non-residents (circle; N=15), residents (triangle; N=31), or unidentified (square; for birds not identified to species level, N=5).

Humanities and Behavioral Sciences [CHBS] on Main street, Student Recreational Center on Jefferson St.; Fig. 2A, B, C).

With these building additions and transformations in mind, we began a multi-year study to investigate BWCs at Radford University. Building on previous findings, we chose to investigate a number of potential landscaping or building features that could influence the location and number of BWCs: window area, building height, and two *in situ* vegetation metrics (total vegetation volume and the Levins index of vertical diversity). Next, we investigated features about the avian community: whether the birds were migratory (non-resident, transitory) or presumed resident species, and whether collisions likely occurred overnight or during daylight hours. We hypothesized that we would detect a greater number of BWCs at our newer buildings that possessed relatively greater window area, and that buildings with greater amounts and diversity of vegetation (which we perceive would reflect in the windows) would result in more BWCs. We further hypothesized that we would find no differences in BWCs between non-resident and resident species, and that most documented collisions would be discovered in the morning hours.

METHODS

Bird-window Surveys

With the contributions from more than 30 Radford University students, we surveyed the perimeter of 15 campus buildings once or twice daily, ideally once in the morning and once in the afternoon. Buildings were selected to represent the full spectrum of building size (height and areal footprint), window area, and landscaped vegetation on campus. We completed surveys from 1 February 2018 through 15 November 2018, and from 7 February 2019 through 5 May 2019. From 6 May through 17 June 2019, we surveyed sporadically on 15 days.

Following the protocol of Hager & Consentino (2014), surveyors walked within 2 m of building edges, scanning for potential bird hits; when a bird was discovered, photos were taken and its location was recorded in UTM. We classified legitimate hits as a cluster (>5) of feathers, partial body fragments, or whole bodies. Live, stunned birds also were also included as legitimate hits. We also collected carcasses opportunistically on campus, even if not collected at the 15 buildings and/or not during set surveys. For this reason, not all BWCs documented were included in every analysis.

Bird Identification

Participating students worked together to identify frozen full bird carcasses to species level, if possible, using standard bird field guides and museum specimens. For identification of partial carcasses and groups of feathers, we relied entirely on comparisons to preserved specimens from the Radford University Biology Department's natural history collection (<https://www.radford.edu/content/csat/home/biology/facilities/natural-history-collection.html>). Although not considered a valid BWC in this study, we collected and identified single feathers or small groups of <5 feathers, and retained them to build a library of known bird artifacts. If unidentifiable specimens contained tissue, they were examined via DNA barcoding analyses (see Paniagua-Ugarte et al., 2019).

Landscape Analyses

We (Powers) calculated total window area (m²) through analysis of architectural drawings of each of the 15 buildings. We (all authors) visited buildings and completed in-person measurements to confirm drawing specifications and remove from calculations windows that were opaquely painted. We determined maximum building height (m) through elevational metrics provided in the architectural drawings.

Following methods similar to Francé & Schnell (2002), we measured vegetation *in situ* at points in ca. 40-m increments, around each building (5–18 points/building). We used a range pole, divided into seven 0.5-m increments (0–0.5 m, 0.5–1.0 m, ... 2.5–3.0 m, >3 m). We focused on vegetation at heights of 3 m or less because other studies reported that vegetation only affected BWCs at lower building floors (e.g., Gelb & Delatacruz, 2009). Standing ca. 1 m from the building facade, we documented a vegetation “hit” in the 0.5-m increment when vegetation was directly touching or within 10 cm of the pole. From these hits, we calculated two vegetation metrics: total vegetation volume (TVV; Mills et al., 1991) and the Levins index of vertical diversity (Levins, 1968). We estimated TVV using the formula:

$$TVV = h/10v$$

where *h* = number of intervals for which we documented vegetation hits, and *v* = total number of intervals (the number of points samples around the building).

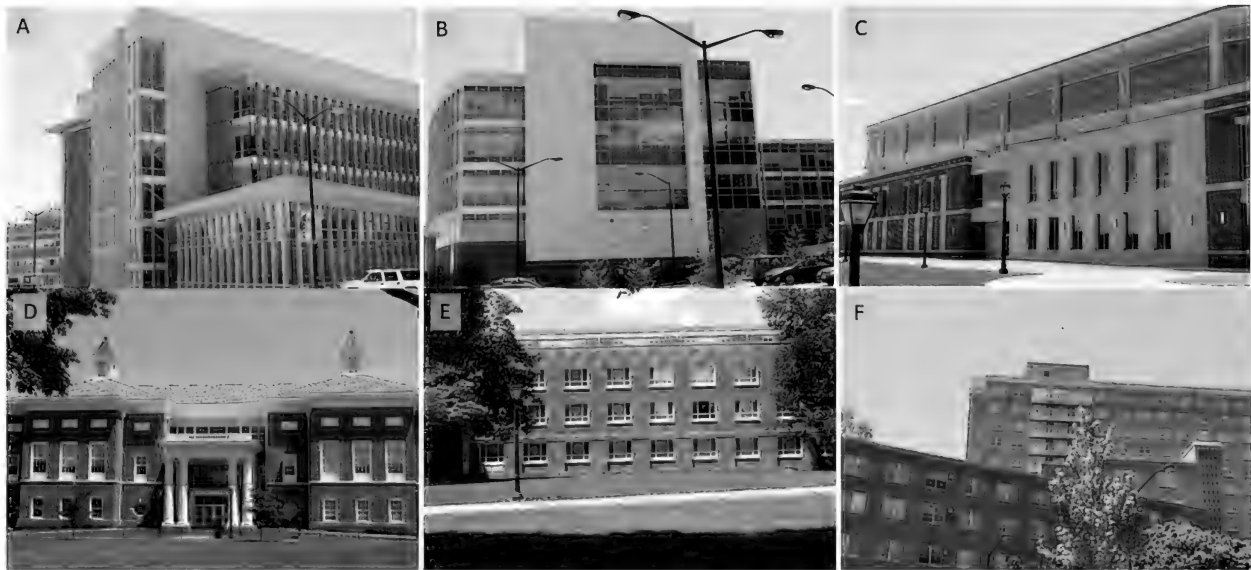


Fig. 2. Examples of surveyed buildings at Radford University, documenting new (<5 years old) buildings on campus that incorporate large window areas (A: Center for the Sciences, B: College of Humanities and Behavioral Sciences [CHBS], C: Student Recreational Center) and more traditional buildings with lesser total window area (D: Whitt Hall, E: Trinkle Hall, F: Muse Hall). Photos by H. Reed, 2019.

The Levins index is defined as: $L = \sum 1 / [(d_i)^2]$

where $d_i = \frac{\text{total number of hits recorded for a 0.5-m increment "i"}}{\text{total number of points measured around the building}}$

Statistical Analyses

We utilized a forward stepwise regression comparing the number of BWC casualties per building to four metrics of each building: total window area, maximum building height, total vegetation volume, and Levins index. Setting a p-value of 0.25 to be included in the model, *a priori*, we ran the regression in JMP Pro 13 (SAS Institute, Cary, NC). We utilized a chi-square goodness of fit test in Microsoft Excel (Microsoft Excel 2019 MSO, Redmond, Washington) to determine if an equal number of carcasses were discovered in the morning (AM) versus the evening (PM) surveys. We considered morning hits as those discovered in daylight surveys completed from ca. 0600–1200 h. Evening hits were those discovered from 1201–1800 h. We recognize that hits that occurred overnight (1801 h–0559 h) are lumped with the morning collections, and we would therefore expect that, if collision patterns were random, we would expect 75% of carcasses to be collected during morning surveys and 25% during evening surveys. Further, we limited our analyses to birds collected on dates in which two surveys/day were completed, so that

we could confidently assign the correct collision time block.

In the same manner, we used a chi-squared goodness of fit test in Microsoft Excel to determine if an equal number of hits occurred for birds considered residents versus those actively migrating (i.e., suspected to collide with buildings while in novel surroundings). Here, we defined “resident” as a bird who is present in the area year-round or migratory but a full-time inhabitant during summer months. These birds would be expected to be familiar with the surroundings. Birds were assigned migratory, non-resident status if they were collected during the species’ known migration period; we assumed the area was unfamiliar to them. Residency status was derived from Cornell Lab of Ornithology’s Birds of North America (Rodewald [Ed.], 2015) using geographic range maps, text, and annual cycle figures (when available). For questionable birds whose migratory status was unclear in southwestern Virginia, we further investigated status utilizing information from the Virginia Breeding Bird Atlas (<https://ebird.org/atlasva>) and Christmas Bird Counts (<https://www.audubon.org/conservation/science/christmas-bird-count>) from the region. Migratory status could only be assigned for ca. six months of the year (3 months for spring migrations, three months for fall migrations) and resident status could be assigned year-round. Therefore, we expected to detect twice as many residents as non-residents by chance alone.

RESULTS

In >975 hours of surveys across 393 days, we documented 51 BWCs across 23 species at Radford University (Appendix 1; Figure 1, 3). BWCs/building ranged from 0–8. Of the 51 birds, one American robin (*Turdus migratorius*) was founded alive but stunned (Fig. 3D); it flew away when the observer attempted to collect it. Sixteen full carcasses (deceased, Fig. 3A, B, C) and 34 partial carcasses or piles of feathers also were collected (Fig. 3E, F). Fifteen individuals across 10 species were non-residents. We found that 31 individuals among 15 species were resident species. Five songbird individuals were not able to be fully identified to the species level, and were not included in this analysis (Fig. 1). A chi-squared goodness-of-fit test for these 46 individuals revealed no significant difference between resident and non-resident species BWC rates ($\chi^2=0.011$, $df=1$, $p=0.917$).

In the 392 days of surveys, we completed 863 individual campus walks. In 827 walks in which time was recorded, 368 were completed in the morning time block and 459 were evening surveys. When we factored out days in which single walks were completed (i.e., we were not able to confidently determine which time block the collision actually occurred), our sample size was reduced to 18 documented collisions. Recording ten hits in the AM block and eight in the PM block, our chi-squared goodness-of-fit test suggested that PM hits occurred marginally more than expected by chance alone ($\chi^2=0.363$, $df=1$, $p=0.056$).

Forty-eight of our 51 BWCs occurred at the 15 buildings for which we calculated window area, building height, and quantified vegetation. Our forward stepwise regression, comparing the number of hits per building versus the four variables reported that the only significant variable was window area ($r^2 = 0.335$, $F = 6.558$, $p=0.024$; Table 1). As window area increased, so did the number of BWCs/building.

DISCUSSION

Our finding that window area was the only metric significantly affecting BWC was not surprising, as the majority of BWC studies have detected this same primary factor across the landscape. Building height may have been less of a factor on this campus because, as originally stated, most buildings are of similar height; however, newer constructions and renovations incorporate markedly more windows into their facades. Perhaps time since construction may have been a co-predictor (with window area) of BWCs on campus, but this metric may not be transferrable to other studies. We also failed to find any vegetation effects on BWCs. Because of the plethora of metrics utilized to measure these features, we either selected metrics that did not accurately account for vegetation around buildings, or we looked at too fine of a scale for vegetation to have affected these birds. Perhaps future studies will rely instead on a broad-scale GIS component, as several studies did find significant, though secondary, effects of vegetation on BWCs (e.g., Hager et al., 2017; Schneider et al., 2018).

The discovery of only 51 bird carcasses in nearly 1,000 h and 393 days of surveys is surprisingly low, compared to other BWC studies across the continent. Locally, Schneider et al. (2018) documented 240 individuals in a shorter time span, only surveying “when schedules and weather allowed.” Our efforts were highest (2 surveys/day) during the fall and spring semesters, which should have corresponded with migratory patterns of birds. We were, at the very least, consistent (1 survey/day) during summer months and when school was not in session. Our efforts attempted to minimize time for scavengers to access the carcasses, yet only 16 (17 if the stunned, live bird is included) full, intact carcasses were discovered. The remaining 34 birds suggested scavenging had occurred (Fig. 3E, F). Nocturnal scavenging events would be expected, as

Table 1. Results of forward stepwise regression, examining factors influencing number of bird-window collisions per building at 15 buildings on Radford University’s campus, 2018–2019. Of four metrics, window area alone explained 33.5% of total variance; no other variables were included in the final model.

Parameter	Estimate	DF	SS	F	p
Intercept	1.6770	1	0	0	1.000
Window area (m ²)	0.0017	1	44.576	6.558	0.023
Building height (max., m)	0	1	1.537	0.212	0.653
Levins	0	1	1.231	0.170	0.688
TVV	0	1	7.535	1.119	0.311

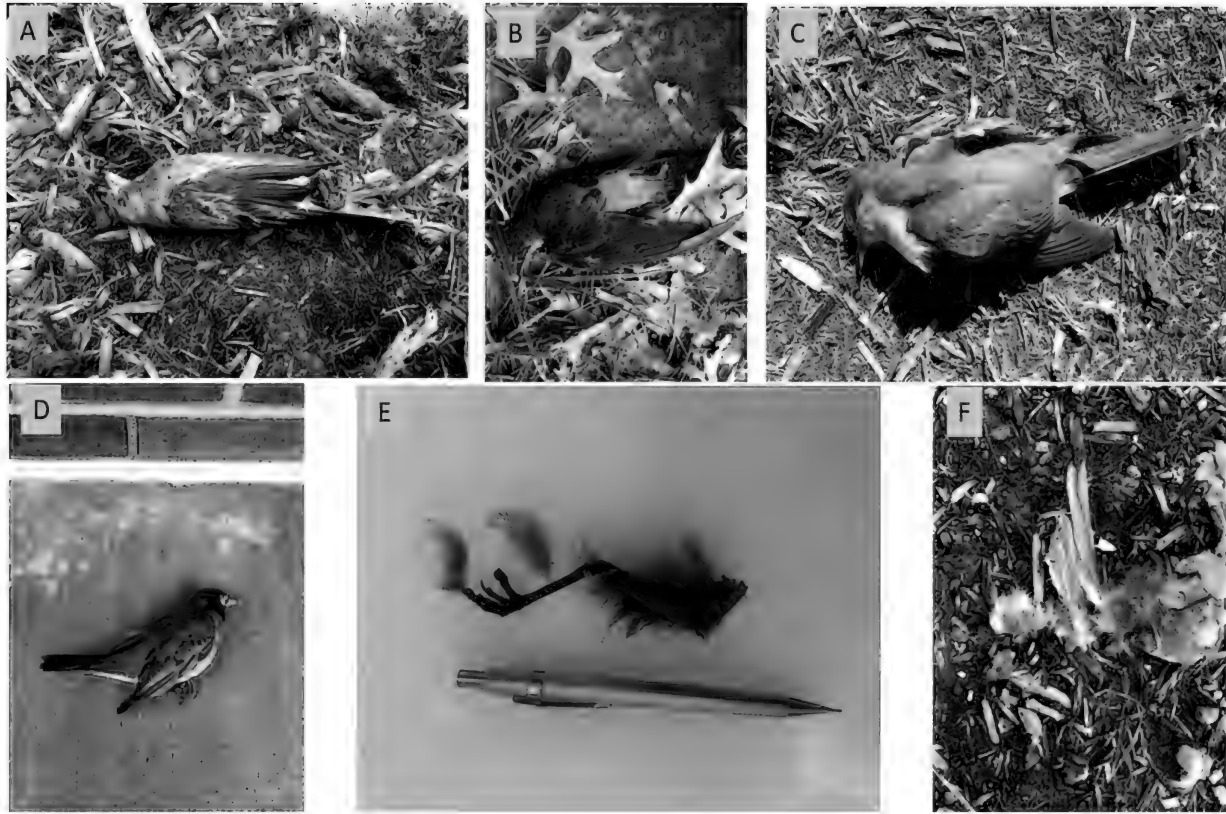


Fig. 3. Examples of casualties from bird-window collisions. Of the 51 documented collisions, 16 were whole bodies - deceased (e.g., A: House Finch [*Haemorrhous mexicanus*], B: Chimney Swift [*Chaetura pelagica*], C: Yellow-billed Cuckoo [*Coccyzus americanus*]), 1 was stunned but recovered (D: American Robin [*Turdus migratorius*]), and the rest were portions of scavenged carcasses (e.g., E: Gray Catbird [*Dumetella carolinensis*], F: White-breasted Nuthatch [*Sitta carolinensis*]). Photos by six participating students at Radford University, 2018-2019.

personal observations include Striped Skunks (*Mephitis mephitis*), Raccoons (*Procyon lotor*), Virginia Opossums (*Didelphis virginiana*), and feral cats as on-campus visitors. Future studies may involve setting a wildlife camera on planted carcasses to determine frequency of and time until documented scavenging or unanticipated anthropogenic disturbances, like students or facilities workers collecting the carcass.

As multiple studies have acknowledged, it is likely that our 51 mortalities are underreporting the actual number of BWCs (Bayne et al., 2012; Kummer et al., 2016b). Besides carcass scavenging (Hager et al., 2012), observer bias plays a significant role in documenting BWCs. With over 30 (albeit trained) students contributing to our project, we assume the visual acuity, mental focus, and ability to detect feathers and partial or full carcasses varied by student (Hager & Cosentino, 2014). In other bird carcass surveys, researchers suggest that the actual number of bird mortalities is 2.3–5 times greater than what is discovered (Dunn, 1993; Zimmerling et al., 2013).

Furthermore, despite the finding from other studies (e.g., Gelb & Delacretaz, 2006; Kahle et al., 2016) that most BWCs occurred during daylight hours, and a marginally significant finding to support that, we are not confident about our sample size. Our intensive twice-daily surveying efforts were too inconsistent across the study, and we could only include 18 of the 51 carcasses for statistical analysis. Our future efforts on campus may investigate short (2–3 week) efforts at buildings with the highest rate of collisions. We might complete three surveys daily, at 8-h intervals, to tease apart collision-time trends. The shorter time frame and subset of buildings might make such studies temporally feasible, given student schedules.

The near-absence of rare or protected species in our observations is interesting. Indeed, none of the 23 species are listed as species of greatest conservation need in the Virginia Wildlife Action Plan (VDGIF, 2015), and none are afforded state-threatened or endangered status. Although we documented three fairly uncommon warblers – Magnolia (*Setophaga magnolia*, 10 October

2018; S2B status – suggesting they are rare breeders in Virginia, Wilson & Tuberville, 2003), Cape May (*S. tigrina*; 29 September 2018), Worm-eating (*Helminthos vermivorum*; 7 May 2018) - all were collected during peak migration periods. Indeed, eBird records document other individuals in the area – some along the New River in Radford – within a two-week window of these finds (<https://ebird.org/atlasva/explore>). Of the 23 species documented, only the Swainson's Thrush (*Catharus ustulatus*) was an unexpected seasonal find. Documented on 30 June 2018, the timing is long after the putative migratory season has concluded; the only Radford record of this species on eBird was on 23 May 2016 (reported by C. Kessler, <https://ebird.org/atlasva/map/swathr>), coinciding with migration periods. Furthermore, just one June record has been reported from nearby counties (Giles Co., VA/Monroe Co., WV line, C. Kessler, pers. comm.). Our mid-summer collision record suggests this individual may have been maintaining a summer residence in the area. This species was identified only by DNA analyses (Paniagua-Ugarte et al., 2019), and we cannot know the age, sex, or any other natural history characteristics of this individual. This species is state-ranked as S1B (Wilson & Tuberville, 2003), suggesting that it is an extremely rare breeder in the Commonwealth. The Virginia Fish and Wildlife Information Service (VaFWIS) system also indicates that this species has not been documented in Radford City in June and that all regional records of Swainson's Thrush were reported during the migratory seasons (S. Watson, VDGIF, pers. comm.). The natural history of this species in our region certainly warrants further investigation.

Twenty-one of our 51 BWCs occurred at only three buildings, all newly-constructed in the last five years (Fig. 2A, B, C) and all possessing substantial window areas (1685-3865 m²). As it appears that Radford University is implementing greater window areas in new construction, we strongly suggest that bird-detering efforts be applied. Window decals can be useful on a small-scale, and even applied on a window-by-window basis by concerned faculty members (as many personal offices contain windows). However, it is unlikely that decals, typically with patterns to make the window more visible to the birds, could or would be utilized on aspects whose window areas comprise nearly 100% of the facade (e.g., Fig. 2A, B, C). The American Bird Conservancy has published a number of window types and the related “threat factor” for BWCs (American Bird Conservancy, 2012). Patterned glass (simple, vertical lines are suggested), translucent glass, and glass coated with UV-reflecting lines all could reduce BWCs, and contribute to LEED-certification (Klem, 2009; Green Building Alliance, 2016). Currently,

bird-detering window modifications offer a pilot credit towards said certification (American Bird Conservancy, 2012). Our project, therefore, provides useful information to the Radford University Office of Planning and Construction, as they design and implement the renovations and new construction on campus. Our data will help the university identify existing areas for potential treatment, as funds become available (M. Biscotte, Radford University Office of Planning and Construction). Implementing such building modifications could establish Radford University as a leader in “green” architecture and provide new research opportunities for students in coming years.

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We are grateful to the more than 30 Radford University students who contributed to BWC surveys. We thank Michael Biscotte and Benny Skeens of the Radford University Office of Planning and Construction providing historical and contemporary architectural drawings of the campus buildings. The Radford University Department of Biology supported this research-based course in bird-window collisions.

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Appendix 1. List of 51 individual birds across 23 confirmed species that were BWC casualties at Radford University in 2018 (February–October) and/or 2019 (February–June). An “*” indicates that one individual was confirmed via genetic analyses of carcass tissue (Paniagua-Ugarte et al., 2019).

Family	Scientific name	Common name	Number of individuals
Order Caprimulgiformes			
Apodidae	<i>Chaetura pelagica</i>	Chimney Swift	1
Order Columbiformes			
Columbidae	<i>Columba livia</i>	Rock Dove	6
Columbidae	<i>Zenaida macroura</i>	Mourning Dove	4
Order Cuculiformes			
Cuculidae	<i>Coccyzus americanus</i>	Yellow-billed Cuckoo	1
Order Passeriformes			
Bombycillidae	<i>Bombycilla cedrorum</i>	Cedar Waxwing*	2
Cardinalidae	<i>Cardinalis cardinalis</i>	Northern Cardinal	1
Certhiidae	<i>Certhia americana</i>	Brown Creeper	1
Corvidae	<i>Corvus brachyrhynchos</i>	American Crow	2
Fringillidae	<i>Haemorhous mexicanus</i>	House Finch	1
Fringillidae	<i>Spinus tristis</i>	American Goldfinch	2
Icteridae	<i>Molothrus ater</i>	Brown-headed Cowbird	1
Mimidae	<i>Dumetella carolinensis</i>	Gray Catbird	2
Paridae	<i>Baeolophus bicolor</i>	Tufted Titmouse	3
Parulidae	<i>Helminthos vermivorum</i>	Worm-eating Warbler	1
Parulidae	<i>Setophaga magnolia</i>	Magnolia Warbler	1
Parulidae	<i>Setophaga tigrina</i>	Cape May Warbler*	1
Passerellidae	<i>Melospiza melodia</i>	Song Sparrow	1
Passerellidae	<i>Spizella passerina</i>	Chipping Sparrow	1
Passeridae	<i>Passer domesticus</i>	House Sparrow*	2
Sittidae	<i>Sitta carolinensis</i>	White-breasted Nuthatch	1
Turdidae	<i>Catharus ustulatus</i>	Swainson's Thrush*	1
Turdidae	<i>Sialia sialis</i>	Eastern Bluebird	2
Turdidae	<i>Turdus migratorius</i>	American Robin	8
		Unknown songbird	5

Using DNA Barcoding to Identify Carcasses from Bird-window Collisions at Radford University

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ABSTRACT

A leading cause of avian mortality is collisions with building windows. To fully understand the impacts of bird-window collisions at Radford University, Virginia, bird carcasses (whole or in part) were collected and documented in 2018–2019. Although the majority of carcasses were identified via morphological features, the taxonomic identification of some samples was impossible due to evidence of predation, environmental degradation, and confusion in species differentiation due to sex, age, and seasonal plumage. We used DNA barcoding to identify carcasses in cases where species identification based on morphology was not possible. DNA barcoding with standard PCR primers allowed for the successful identification of five individuals across four species - two of which species had not been previously identified in this study. Our study emphasizes the application of DNA barcoding in bird-window collision studies, and its potential for use in other conservation and mitigation efforts.

Keywords: avian mortality, cytochrome c oxidase subunit 1, species identification, taxonomy.

INTRODUCTION

Bird-window collisions (BWCs) are a leading cause of mortality in the United States and world-wide, with annual mortality estimates of nearly one billion birds (Loss et al., 2014). Such collisions are suspected to be greater during peak migratory periods, as birds traverse less familiar habitats (Borden et al., 2010; Schneider et al., 2018). Borden et al. (2010) reported that migratory birds were nine times more likely to be a casualty of BWCs than resident species. While some studies report that particular species or family groups are more prone to collisions (e.g., hummingbirds, Schneider et al., 2018), it is evident that both common and rare species are susceptible to this threat.

Multiple large-scale and/or long-term studies of BWCs acknowledge that mortalities are likely under-reported (Bayne et al., 2012; Kummer et al., 2016). A major limitation is observer error, which manifests itself as a failure to detect carcasses that are present (e.g., carcasses obscured by vegetation). Carcass scavenging is a second major cause of BWC underestimates (e.g., Kummer et al., 2016). Finally, carcasses landing outside a limited search area may also affect discovery

(Zimmerling et al., 2013). Recent estimates suggest that carcass recovery is 2.3–5 times less than the actual number of bird mortalities (Dunn, 1993; Zimmerling et al., 2013).

Identification is important because all bird samples collectively play a role in understanding how species migration, seasonal distribution, and density could relate to bird building collisions (Schneider et al., 2018). However, once recovered, several circumstances may limit accurate identification of the carcass. Damage to the specimen resulting from the collision, length of time between mortality and collection, and scavenger activity may all affect the ability to identify that carcass. Additionally, differentiation between juvenile and adult females may be difficult, particularly among passerines. In our BWC study (Powers et al., this volume), all of these factors impeded accurate identification of some specimens.

From February 2018 – June 2019, students at Radford University investigated myriad aspects of natural and anthropogenic influences on the number and nature of BWCs at 15 buildings on the university campus (Powers et al., this volume). Statistical analyses required the accurate identification of bird carcasses. Students

identified full carcasses, partial carcasses, and multiple feather evidence from BWCs via comparisons to bird specimens within the natural history collection at Radford University. However, morphological identification of nine of 51 birds was not possible or definitive.

DNA barcoding, identifying species by comparing a short, defined DNA sequence to a DNA sequence reference database, provided an alternative approach to identifying a specimen (Kerr et al., 2007). DNA barcoding has been effective for the species identification of whole birds (Herbert et al., 2004), bird tissue (Dove et al., 2008), and eggs (Lee & Prys-Jones, 2008) from unknown samples. Of the nine unidentifiable carcasses, seven contained tissue, and thus had the potential to be identified through DNA barcoding. Our goal was to extract DNA from carcasses and use DNA barcoding to identify them to the fullest extent possible.

METHODS

Seven bird carcasses that contained tissue but were physically unidentifiable were collected and stored in a standard chest freezer at 20° C. One additional BWC specimen collected in October 2016 at Radford University was included in this project, increasing our sample size to eight.

DNA was extracted from tissues using the Qiagen DNeasy blood and tissue kit (Qiagen, Austin, Texas). We amplified a 708 bp fragment of the mitochondrial encoded cytochrome c oxidase subunit I (COI) gene using tailed primers BirdF1_t1 and BirdR1_t1 (Kerr et al., 2007). Each 50 µl PCR reaction contained 25 µl of 2x Quick-Load Taq master mix (NEB, Ipswich, Massachusetts) 2.3 mM MgCl₂, 0.5 mM each of forward and reverse primers, 5 µl of DNA template (approx. 200 ng), and distilled water. PCR amplification was performed with an initial denaturation (3 min at 94° C) followed by 35 cycles of 94° C for 1 min, 51° C for 1 min, and 68° C for 1 min. This was followed by a final extension step at 68° C for 10 min. PCR reactions were held at 4° C for 6 to 8 h until storage at -20° C.

We identified successful amplification by running 5 µl of each sample on 2% agarose/TBE/EtBr gel. Successfully amplified samples were sent to GENEWIZ (South Plainfield, New Jersey) for sequencing. Both strands of our amplicons were sequenced using the Sanger dideoxy chain termination method (Sanger & Coulson, 1975). We used the DNA subway (DNA Learning Center, <https://dnasubway.cyverse.org>) as a means to align forward and reverse sequences of each sample and to evaluate/correct discrepancies between the sequences of the forward and reverse strands and to construct a consensus sequence of the forward and

reverse strands. We used the “Identification Engine” (http://boldsystems.org/index.php/IDS_OpenIdEngine) to identify the taxonomic origin of unknown specimen DNA using the consensus sequence as a query. We assume “correct” species identification for samples that show ≥99.0% sequence similarity with bird species already in the database.

RESULTS

Of the eight carcasses tested, we successfully amplified and identified five to species level (Table 1). We confidently identified one Cedar Waxwing (*Bombycilla cedrorum*), one House Sparrow (*Passer domesticus*), two Cape May Warblers (*Setophaga tigrina*; one was the 2016 collection), and one Swainson’s Thrush (*Catharus ustulatus*). The thrush and the Cape May Warblers had not been previously identified in the BWC project. The specimens whose DNA did not amplify remain unidentified via morphological features.


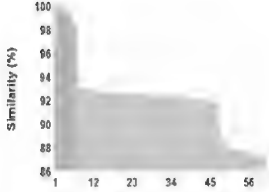
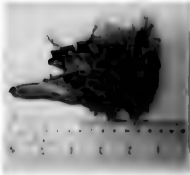
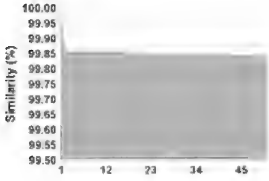
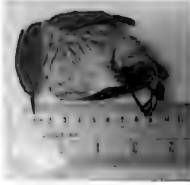
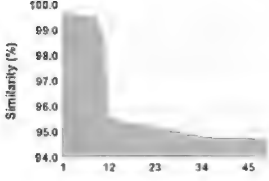

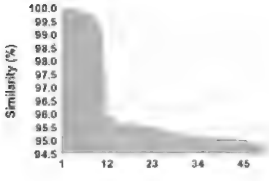
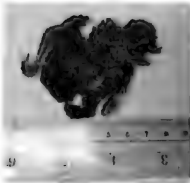
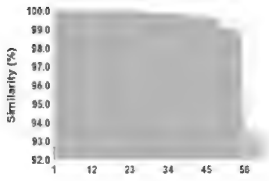
DISCUSSION

The use of DNA barcoding allowed for the identification of five of the eight unidentifiable bird carcasses. All five of these carcasses had a sequence similarity of ≥99.0% to reference species and are considered to be accurately identified. Unsuccessful amplification may be due to the degradation of DNA that results with age and/or the environmental conditions experienced before collection. There are, however, various methods that may allow for the analysis of degraded DNA such as the PCR amplification of shorter sequences using internal primers.

While we used primers designed to amplify the complete 708 bp “Folmer region” of the COI gene (Folmer et al., 1994), these primers prove ineffective at amplifying all samples. Often, this results from the fragmentation of DNA due to environmental degradation which results in the lack of a full-length DNA template. Future research will be aimed at designing internal primers which will allow the amplification of shorter target sequences and/or the application of DNA mini-barcodes (Meusnier et al., 2008) to these problematic specimens. This should allow us to apply DNA barcoding to a more degraded template.

The results from the five successfully identified samples provide useful data to help us understand the relationships of particular bird species with BWCs. Of greatest interest were the two species (Swainson’s Thrush and Cape May Warbler) that had not previously been identified as BWC casualties in this project. Both are migratory species in the region and not considered

Table 1. Bird-window collision carcasses collected from Radford University (2016, 2018-2019) that were identified with DNA barcoding. Presented is a photo of each original carcass, the range of sequence similarity to members of the top matched species, and the DNA percent match, broken down by family, genus, and specific epithet. All samples fall into the order Passeriformes.

SAMPLE	TAXON ASSIGNMENT	PROBABILITY OF PLACEMENT (%)	SEQUENCE SIMILARITY TO TOP 100 DATABASE MATCHES
	Bombycillidae <i>Bombycilla</i> <i>cedrorum</i> Cedar Waxwing	100 100 100	
	Passeridae <i>Passer</i> <i>domesticus</i> House Sparrow	100 100 100	
	Parulidae <i>Setophaga</i> <i>tigrina</i> Cape May Warbler	100 100 99.7	
	Parulidae <i>Setophaga</i> <i>tigrina</i> Cape May Warbler	100 100 100	
	Turdidae <i>Catharus</i> <i>ustulatus</i> Swainson's Thrush	100 100 100	

summer residents. Radford University's campus abuts the New River, a migratory pathway. Therefore, it is not entirely unexpected to find these species. While both species are uncommon, they have been documented along the New River in Radford. The Cape May Warbler has been observed in late September and early October, and the Swainson's Thrush has been observed most often in September through mid-October with sporadic observations in May (e.g., eBird data for Bisset Park and Riverway Trail, Radford, Virginia; <https://ebird.org/atlasva>). The Swainson's Thrush was collected on 30 June 2018, providing the first local record of this taxon outside regular migration periods. To date, only one June record in the region has been reported (Giles Co., VA/Monroe Co., WV line, C. Kessler, pers. comm.). Powers et al. (2019) review these species in greater detail. The identification of the Swainson's Thrush demonstrates the power of DNA barcoding as this potentially significant observation would have otherwise gone unnoticed.

We have demonstrated the power of DNA barcoding in providing an alternative means for the taxonomic identification of specimens where, for numerous and varied reasons, traditional means of identification based on morphology may be inconclusive or impossible. While we have applied this approach to the identification of avian taxa, it is easy to see where this approach would be useful in other studies as well. DNA barcoding could easily be applied to the taxonomic identification of tissues from roadkills, prey remains found in association with predatory animals, and cryptic species where the identification based on morphological differences is problematic. These methods could be particularly useful in regulatory projects, like surveys around wind turbines (for birds and bats), where identification to species level may have greater implications for mitigation efforts.

Applying DNA barcoding as an approach for species identification should not fall outside the realm of many studies. With the exception of DNA sequencing, the molecular techniques necessary to complete this project are common among introductory biology and genetics college-level courses. Myriad of companies are available to perform the Sanger dideoxy sequencing reaction if these are unavailable in-house. The cost per sample of DNA barcoding can range from between \$5.00 to \$10.00 USD depending upon: in-house versus commercial sequencing, sequencing one or both strands or, the necessity for band isolation to remove primer dimers. If one is distinguishing among only a few potential taxa, a related approach, Cleaved Amplified Polymorphic Sequences (CAPS; Konieczny & Ausubel, 1993) or the related dCAPS (derived Cleaved Amplified Polymorphic Sequences; Neff et al., 1998) could

successfully be applied to species identification saving time and reducing the cost of analysis to a few dollars.

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<http://dx.doi.org/10.5751/ACE-00609-080210>

The Rove Beetles (Coleoptera: Staphylinidae) of the George Washington Memorial Parkway, with a Checklist of Regional Species

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ABSTRACT

Two-hundred and nine taxa (171 identified to species level), in 111 genera, of staphylinid beetles were documented during a 21-year field survey of a national park site (George Washington Memorial Parkway) that spans parts of Fairfax and Arlington counties in Virginia. Fifty-two species, plus five additional genera, documented from the Parkway are first records for Virginia. An additional 62 species new to Virginia are listed in the appendix from broader research documenting 792 species of Staphylinidae from Virginia, Maryland, and the District of Columbia. The study also increases the number of staphylinid species known from the Potomac River Gorge to 167. Of the seven capture methods used in the survey, Malaise traps were the most successful. Periods of adult activity, based on dates of capture, are given for each species. Relative abundance is noted for each species based on the number of captures. Notes on morphological characteristics and habitats are given for some species. Thirteen species adventive to North America are documented from the Parkway and 60 adventive species are recorded from Virginia, Maryland, and the District of Columbia in the Appendix. Range extensions are documented for 16 species. Images of 11 species new to Virginia are provided.

Keywords: Biodiversity, District of Columbia, insects, Malaise traps, Maryland, national parks, new state records, Potomac River Gorge, Virginia.

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INTRODUCTION

Probably the largest family of beetles in the world, the Staphylinidae, or rove beetles, contains more than 64,031 species in 32 subfamilies and 167 tribes (Irmeler et al., 2018; Newton, 2019). It is the largest beetle family in North America, with 568 genera containing over 4,500 species (Newton et al., 2000; Newton, 2019). They are generally recognized by their short, truncate elytra that leave exposed a large dorsal part of the abdomen. In many tribes the abdomen is flexible and is wagged from side to side as the beetle moves along the ground. In a few groups the abdomen is less flexible (Micropeplinae) or the elytra cover the abdomen (some Omaliinae, Scydmaeninae). Possible staphylinid fossils have been found near the Jurassic/Triassic boundary that are more than 200 million years old, and numerous subfamilies were present by the end of the Jurassic (Newton, et al., 2000; Chatzimanolis, 2018).

No other group of Coleoptera has been as successful as the Staphylinidae at living in such an enormous number of diverse habitats (Thayer, 2016; Betz et al., 2018). Adults of most species are nocturnal or shelter in dark areas during daylight hours; some exceptions include *Stenus* species, which are active in daylight. They are common components of soil biota, found in decaying leaf litter or deeper soil layers where they prey upon a variety of soil inhabiting organisms, or feed on decaying plant material or on fungi. However, this family fills nearly every ecological niche and they can be found in fungi, rotting wood, dung, carrion, caves, mammal burrows, and ant or termite nests. Some smaller species (most species are 2–8 mm) have an ant-like habitus and some species (especially Aleocharinae) live among particular ants or other social Hymenoptera or termites. Others are associated with birds or ectodermally on mammals (Brunke & Buffam, 2018). These associations can be beneficial (consuming the host waste material) or parasitic, when they prey on the eggs, larvae or stored food of the hosts. The larvae of *Aleochara* are parasitoids of dipterous puparia. Because of their hyper-diversity of form, habitats, and prey, their ancient origins, and relatively well-preserved fossil history, the Staphylinidae make interesting subjects for scientific study and they are becoming more widely used as bioindicators of environmental conditions in applied sciences such as forest research and conservation (Klimaszewski et al., 2018). A few species are even of medical importance (e.g., some *Paederus* spp.) or have been proposed or used in biological control (e.g., *Aleochara* spp.) (Thayer, 2016). This study adds to our knowledge of the distribution and life histories of the Staphylinidae of the Mid-Atlantic area of North America.

STUDY SITES

The study site includes lands managed by the National Park Service as units of the George Washington Memorial Parkway (GWMP) in Virginia (Fairfax and Arlington counties). Park sites that received the greatest inventory effort included: in Fairfax County, Dyke Marsh Wildlife Preserve, Fort Marcy, Great Falls Park, Little Hunting Creek, and Turkey Run Park and in Arlington County, Arlington Woods (at Arlington House), Gravelly Point Park, and Gulf Branch.

A map of these sites is provided in Steury (2011). This area covers approximately 1,615 ha. Great Falls and Turkey Run parks and Fort Marcy and Gulf Branch fall within the Piedmont physiographic province while all other collection sites are on the Coastal Plain. Most sites are situated along the shore of the Potomac River, and the Piedmont sites border the Potomac River Gorge, an area known for high species richness of plants and animals (Cohn, 2004). Most of the study sites are dominated by maturing, second growth, primarily upland, deciduous woodlands.

However, older-age stands, with dominant trees over 100 years old, occur on ridges at both the northern and southern ends of Great Falls Park. Abrams & Copenheaver (1999) documented White Oak (*Quercus alba* L.) individuals between 208 and 251 years old and a Black Gum (*Nyssa sylvatica* Marshall) 166 years old along the northern ridge. Counts of radial growth rings in 1994 on a Shortleaf Pine (*Pinus echinata* Mill.) that fell from a ridge along the southeastern edge of the Park dated to at least 220 years old (R. Simmons pers. comm. 2007). More open habitats can be found in moist, narrow, herbaceous dominated bands along the shore of Potomac River, in the freshwater, tidal, swamp and marsh habitats at Dyke Marsh, and in areas with managed turf grass and scattered large trees, such as Collingwood Picnic Area and Gravelly Point Park. The vascular flora of the GWMP is diverse, with more than 1,313 taxa recorded, 1,020 from Great Falls Park alone (Steury et al., 2008; Steury, 2011).

MATERIALS AND METHODS

Specimens were collected during a 21-year period (1998–2018) using a variety of sporadic survey methods targeting arthropods, including: Malaise traps, Lindgren funnels, blacklight (UV) bucket traps, blacklight shone on sheets, leaf litter samples processed in Berlese funnels, pitfall traps, and hand picking (including splashing along river shoreline). Six Townes style Malaise traps (Townes, 1962) were set at Dyke Marsh, April 1998–December 1999, three each at Great Falls and Turkey Run parks (March 2006–December 2009), and

four at Little Hunting Creek (March–November 2017 and 2018). Traps at Dyke Marsh were set each year in the same locations in open, tidal, freshwater marsh dominated by *Typha angustifolia* L.; floodplain forest dominated by Red and Silver maple (*Acer rubrum* L. and *A. saccharinum* L.) and Tulip Poplar (*Liriodendron tulipifera* L.); and at the marsh/forest ecotone. In Great Falls Park, a trap was set in each of three habitats: quarry site (dry, upland, mixed deciduous/coniferous forest), swamp (dominated by Red Maple), and floodplain forest (dominated by oaks [*Quercus* sp.], and Tulip Poplar). In Turkey Run Park, one trap was set in upland forest dominated by oaks and tulip poplar and two traps in floodplain forest along the Potomac River (dominated by oaks, Basswood [*Tilia americana* L.], and Sycamore [*Platanus occidentalis* L.]). At Little Hunting Creek, four traps were set in upland forest dominated by an ericaceous understory and a canopy of oaks, hickory (*Carya* sp.), American Beech (*Fagus grandifolia* Ehrh.), and some Virginia Pine (*Pinus virginiana* Mill.).

Additional collections were also made by sporadically using other collecting methods, including running pitfall traps set at Dyke Marsh (five years), at Little Hunting Creek, and Great Falls and Turkey Run parks (three years), and Arlington Woods and Gulf Branch (two years); Lindgren funnel and blacklight (UV) bucket traps set at Dyke Marsh, Great Falls Park, Little Hunting Creek, and Turkey Run Park (two years); blacklight shone on sheets at Great Falls and Turkey Run parks (three years); leaf litter from Arlington Woods, Dyke Marsh, Fort Marcy, Gravelly Point Park, Great Falls Park, Gulf Branch and Turkey Run Park, processed in Berlese funnels (two years) and collecting by hand at all sites, over seven years. Hand collecting was aided by splashing (pouring water over shoreline and gravel bar habitats) unless otherwise indicated in the list of species. Locations, habitat descriptions, and collection methods are summarized in Table 1. Individuals who collected specimens of staphylinid beetles from GWMP included C. Acosta, E. Barrows, J. Brown, C. Davis, A. Evans,

Table 1. Summary of locations, latitude and longitude, habitats sampled, and trap types used during this study. Additionally, all sites were sampled by hand picking (hp), often aided by splashing water along river shoreline.

Location	Latitude/ Longitude	Habitats Sampled	Trap or Collection Types
Arlington County			
Arlington Woods (AW)	38.883 -77.074	Upland, deciduous forest	Berlese funnels (bf), pitfall traps (pf)
Gravelly Point Park (GP)	38.864 -77.039	Turf grass and river shoreline	Berlese funnels
Gulf Branch (GB)	38.924 -77.115	Upland, deciduous forest along stream	Berlese funnels, pitfall traps
Fairfax County			
Dyke Marsh Wildlife Preserve (DM)	38.772 -77.050	Tidal, freshwater marsh; floodplain forest; marsh/forest ecotone	Berlese funnels; black-light (UV) bucket traps (bt); Lindgren funnels (lf); pitfall traps; Townes style Malaise traps (mt)
Fort Marcy (FM)	38.937 -77.125	Upland, deciduous forest	Berlese funnels
Great Falls Park (GF)	38.985 -77.246	Upland, dry, mixed deciduous/coniferous forest; deciduous swamp; deciduous floodplain forest	Berlese funnels; black-light shone on sheets (bl); black-light (UV) bucket traps; Lindgren funnels; pitfall traps; Townes style Malaise traps
Little Hunting Creek (LH)	38.717 -77.078	Upland deciduous forest with some pine	Black-light (UV) bucket traps; Lindgren funnels; pitfall traps; Townes style Malaise traps
Turkey Run Park (TR)	38.965 -77.156	Upland deciduous forest; floodplain deciduous forest	Berlese funnels; black-light shone on sheets; black-light (UV) bucket traps; Lindgren funnels; pitfall traps; Townes style Malaise traps

J. Fisher, S. Lingafelter, D. Mead, E. Oberg, M. Skvarla, D. Smith, W. Steiner, B. Steury, J. Swearingen, and C. Wirth.

Specimens were determined by R. M. Brattain, A. J. Brunke, D. S. Chandler, A. V. Evans, M. L. Ferro, C. Francois, C. W. Harden, E. R. Hoebeke, J. E. Louderman, A. Marsh, A. F. Newton, B. W. Steury, and M. K. Thayer. Identifications of taxa outside the specialties of the various determiners were made with the assistance of general identification guides to all or part of North American Staphylinidae, including Downie & Arnett (1996), Newton et al. (2000), Klimaszewski et al. (2018), and Smetana (1995), supplemented by generic revisions such as Campbell (1976, 1982) and many other revisions or published notes, most of which are listed under the relevant genus in Newton et al. (2000). In addition, when possible, identifications were made or confirmed by comparison of GWMP specimens with specialist-identified specimens in the Field Museum of Natural History. In spite of these considerable efforts, the current state of knowledge of staphylinid taxonomy in North America, where many subfamilies and genera are still in need of modern revision and/or lack specialists who can identify them, has precluded the complete identification of the GWMP specimens, with the result that some species could be identified reliably only to genus (indicated as [Genus] “sp.” in the list below). New state record determinations are based on examination of the collections and the literature given in the Appendix. Specimens from GWMP were pinned, labeled, and deposited in the collections maintained at the GWMP, Turkey Run Park Headquarters in McLean, Virginia. Determiners and collection depositories for new state records are given in the Appendix.

RESULTS AND DISCUSSION

Two-hundred and nine taxa (171 identified to species level), in 111 genera, of staphylinid beetles were documented from GWMP. The most species rich subfamilies were Staphylininae (47 taxa), Tachyporinae (33), and Aleocharinae (29). The tribes with the most taxa were Staphylinini (43), Tachyporini (20), and Mycetoporini (13). The most species-rich genera were *Philonthus* (14), *Sepedophilus* (10) and *Lordithon*, *Platydracus*, and *Stenus* (7 each). The most abundant species collected in the survey area were *Philonthus asper* (51), *Platydracus maculosus* (36), *Sepedophilus crassus* (33), *Cyparium concolor* (30), *Tachinus fimbriatus* (27), *Bryoporus rufescens* (26), *Achenomorphus corticinus* (24) and *Oxybleptes kiteleyi* (23). In order to determine which species documented from GWMP are new to Virginia a checklist was created based on literature reviews and searches of multiple

North American collections (see appendix) documenting 558 species from Virginia. Based on the appendix, 52 species, plus five additional genera, documented from the GWMP are first records for Virginia. Additionally, 62 species new to Virginia were documented in other collections (see appendix), rendering a total of 114 species and 34 genera new to Virginia. Range extensions are documented for 16 species (see list of species for details). Of the 167 taxa documented from Piedmont sites along the Potomac River Gorge at Great Falls or Turkey Run parks or Fort Marcy or Gulf Branch during this study, 164 are first records for the gorge (Brown, 2008). *Cyparium flavipes* LeConte and *Scaphidium oblitteratum* LeConte reported from the Potomac Gorge by Brown (2008) have been synonymized with other species documented from the gorge. The GWMP sites with the highest species richness were Turkey Run Park with 125 taxa, Great Falls Park (106), and Dyke Marsh Wildlife Preserve (66). Thirteen species found in GWMP are considered adventive (non-native) to Virginia and 60 species are adventive to the area comprising Virginia, Maryland, and the District of Columbia (see Appendix). Malaise traps proved to be the most successful method of capturing staphylinid beetles during this study, yielding 150 taxa. Comparable figures for other sampling methods were: hand picking (including the use of splashing), 69; Berlese funnels, 40; and pitfall traps, 24. Despite 21 years of sporadic survey effort using seven collecting techniques, 78 taxa (37.3%) documented by this study are represented by a single specimen, indicating that the list of GWMP staphylinids is very preliminary and much remains to be learned concerning the fauna of the parkway and of Virginia.

LIST OF SPECIES

Genera and species are listed alphabetically by subfamily, and then by supertribe, tribe and subtribe (when these are used within a subfamily). An exclamation mark is used to mark taxa not previously documented in Virginia. Adventive (non-native) species are marked with a dagger (†). The number of specimens in the collection at GWMP is indicated in parentheses after each taxon. Collection sites and methods are given using the abbreviations listed in Table 1. Other locations or collection methods are spelled out if necessary. The periods of adult activity are based on dates when live individuals were captured in the park. Dates separated by a hyphen indicate that the taxon was documented on at least one day during each month within this continuum of months, whereas dates separated by a comma represent individual observation dates. For traps set over multiple weeks, the first day of the set is used as the earliest date and the last day of the set as the latest date.

LIST OF SPECIES

Family Staphylinidae

Subfamily Aleocharinae

Tribe Aleocharini

Subtribe Aleocharina

- † *Aleochara lata* Gravenhorst – (4); DM, GF, TR; 11 Apr-8 Aug; mt, pf.
Aleochara lustrica Say – (6); DM, GF, TR; 3 Jul-4 Sep; mt.
 ! *Aleochara ocularis* Klimaszewski – (2); GF; 10-30 Apr; mt. This record documents a southeastern range extension from Pennsylvania and Kentucky.
 ! *Aleochara rubripes* Blatchley – (1); TR; 1-20 May; mt. This specimen represents a southeastern range extension from Pennsylvania and Kentucky.
 ! *Aleochara verna* Say – (2); DM, GF; 13-20 Jun; bee bowl, mt. Fig. 1.

Tribe Athetini

Subtribe Athetina

- Atheta* sp. – (3); DM, LH, TR; 26 May-7 Jul; hp, mt, pf.
 ! *Strigota* cf. *ambigua* (Erichson) – (1); LH; 10 Jun; hp. This is the first record of this genus from Virginia, Maryland, or the District of Columbia.

Subtribe Athetini incertae sedis

- ! *Apalonia seticornis* Casey – (1); AW; 14 May; bf. This record represents a northeastern range extension from Florida and Kansas.

Tribe Falagriini

- Aleodorus bilobatus* Say – (1); TR; 5 Sep-21 Oct; mt.
Borboropora quadriceps (LeConte, J. L.) – (1); TR; 7-21 Jun; mt.
Myrmecocephalus cingulatus (LeConte, J. L.) – (2); DM, TR; 6-20 Jun, 18 Aug-4 Sep; mt.
Myrmecocephalus concinnus (Erichson) – (1); DM; 20 Jun-1 Jul; mt.

Tribe Geostibini

- Aloconota* sp. – (3); GF, TR; 21 May-17 Jul; mt.
Geostiba sp. – (1); FM; 15 Apr; bf.

Tribe Homalotini

Subtribe Bolitocharina

- Leptusa* sp. – (2); GF, TR; 5 Sep-1 Dec; mt.
 ! *Phymatura* cf. *blanchardi* (Casey) – (4); GF, TR; 9 Sep-1 Dec; mt. This is the first record of this genus recorded from Virginia, Maryland, or the District of Columbia. This *Phymatura* species is difficult to identify but is most likely *blanchardi* when compared to previously named museum specimens and referenced to available literature.

Subtribe Gyrophaenina

- Gyrophaena* sp. – (3); DM, GF; 18 May-30 Jul; hp, mt.
Phanerota fasciata (Say) – (2); GF, TR; 19 Jun-17 Jul; mt

Tribe Hoplandriini

Subtribe Hoplandriina

- Hoplandria laevicollis* (Notman) – (3); TR; 19-30 Jun; mt.
Hoplandria sp. – (6); GF, TR; 21 May-30 Jul; mt, pf.

Tribe Hypocyphtini

- Oligota* cf. *pusillima* (Gravenhorst) – (1); GP; 14 May; bf. If identified correctly, this species is believed to be adventive in North America, known from Canada (New Brunswick) and the United States (Massachusetts and New York).

Tribe Lomechusini

Subtribe Myrmedoniina

- Pella* sp. – (3); DM; 15 Apr-18 May; hp.

Tribe Myllaenini

- Myllaena* sp. – (2); DM, TR; 20 Jun-4 Sep; mt.

Tribe Oxypodini

Subtribe Oxypodina

- ! *Gennadota* sp. – (3); TR; 9 Sep-17 Nov; mt. This is the first record of this genus from Virginia, Maryland, or the District of Columbia.

Oxypoda cf. *opaca* (Gravenhorst) – (3); DM, TR; 21 Jul-4 Aug, 21 Nov-31 Jan; mt. Even when compared to previously named museum specimens and referenced to available literature, this species is difficult to identify but is most closely allied to *O. opaca*.

Oxypoda sp. – (2); AW, DM; 14-18 May; bf, hp.

! *Tetralaucopora* sp. – (1); DM; 7-21 Nov; mt. This is the first record of this genus from Virginia, Maryland, or the District of Columbia.

Tribe Placusini

Placusa sp. – (7); DM, TR; 6 Jun-4 Sep; mt.

Tribe Tachyusini

Gnypeta cf. *nigrella* (LeConte, J. L.) – (1♀); TR; 14 Jun; hp. This species is difficult to identify without male genitalia, but is probably *G. nigrella* when compared to previously named museum specimens and referenced to available literature.

Tachyusa sp. – (2); GF, TR; 4-18 May; hp.

Subfamily Euaesthetinae

Tribe Euaesthetini

Euaesthetus cf. *americanus* Erichson – (3); DM; 15 Apr-18 May; hp. This *Euaesthetus* is difficult to identify but is most closely allied to *E. americanus* when compared to previously named museum specimens.

Euaesthetus iripennis Casey – (1); AW, 15 Apr, bf.

Subfamily Megalopsidiinae

Megalopinus caelatus (Gravenhorst) – (2); GF, LH; 2-30 Jun; mt.

Megalopinus rufipes (Motschulsky) – (2); GF, LH; 13 Apr-15 May; mt.

Subfamily Omaliinae

Tribe Anthophagini

Acidota subcarinata Erichson – (1); TR; 22 Oct-1 Dec; mt.

Arpedium schwarzi Fauvel – (7); DM, GF, TR; 15 Apr-7 Jul, 22 Oct-1 Dec; hp, mt, pf.

Arpedium n. sp. – (1); DM; 24 Oct-8 Nov; mt. Fig. 2. Recognized as undescribed by Margaret Thayer based on male genitalia, this new species is also known from North Carolina, Ohio, and Oklahoma. It is being described by Alexey Shavrin in a revision of North American species of *Arpedium*.

Geodromicus brunneus (Say) – (13); GF, LH, TR; 24 Apr-25 May, 22 Oct-1 Dec; bl, hp, mt.

Lesteva pallipes LeConte, J. L. – (6); DM, GF, TR; 27 Apr-20 May, 22 Oct-1 Dec; lf, mt.

Olophrum obtectum Erichson – (6); AW, DM, TR; 18 Mar-14 May, 21 Nov-5 Dec; bf, mt.

Trigonodemus striatus LeConte, J. L. – (3); GF, TR; 5 Sep-17 Nov; mt. This species was first reported from Virginia by Steury (2017).

Tribe Omaliini

Hapalaraea hamata (Fauvel) – (1); TR; 5 Sep-21 Oct; mt.

! *Omaliium* (sensu lato) *fractum* Fauvel – (1); TR; 22 Oct-1 Dec; mt. Fig. 3. (generic placement: see Newton et al., 2000: 338, under *Pycnoglypta*). This first Virginia record fills a gap in the broad distribution extending northwest to Illinois and Michigan; south-southwest to Texas and Oklahoma, Georgia, Kentucky, and North Carolina and northeast to New York.

! *Omaliium repandum* Erichson – (1); TR; 22 Oct-17 Nov; mt. This specimen fills a range gap in a broad distribution extending northeast, west, and northwest.

! *Phyllodrepa humerosa* (Fauvel) – (2); DM, GF; 27 Oct-5 Dec; mt. These Virginia records fill a gap in the broadly documented distribution of this species which extends west to Wisconsin through Oklahoma, south to Georgia, and north to Pennsylvania and Nova Scotia, Canada.

Phyllodrepa punctiventris (Fauvel) – (2); AW; 14 May; bf.

! † *Xylodromus concinnus* (Marshall) – (1); GF; Apr 11-28; lf. This first Virginia record adds to a poorly documented distribution scattered over North America.

Subfamily Osoriinae

Tribe Eleusinini

! *Eleusis pallida* (LeConte, J. L.) – (1); TR; 1-20 May; mt.

Tribe Osoriini

Molosoma latipes (Gravenhorst) – (1); TR; 13 Jun; hp.

Tribe Thoracophorini

Subtribe Clavilispinina

! *Clavilispinus rufescens* (LeConte, J. L.) – (2); AW, LH; 13 Apr-15 May; bf, mt. Fig. 4. These records document a northern range extension from South Carolina.

Subtribe Thoracophorina

Thoracophorus costalis (Erichson) – (2); AW; 15 Apr-14 May; bf.

Subfamily Oxyporinae

Oxyporus stygicus Say – (1); TR; 1-20 May; mt.

Pseudoxyporus lateralis (Gravenhorst) – (1); TR; 21 Jul-4 Aug; mt.

Pseudoxyporus occipitalis (Fauvel) – (1); TR; 1-20 May; mt.

Pseudoxyporus quinquemaculatus (LeConte, J.L.) – (17); GF; 21 May-30 Jun; mt. Twelve of these were captured in the same Malaise trap sample set in Great Falls Swamp.

Subfamily Oxytelinae

Tribe Blediini

Bledius cf. *annularis* LeConte, J. L. – (1); TR; 4 May; hp. More than 28 species of this genus are reported from the Northeast. Some of the distinguishing characters are subtle in structure, making the species difficult to identify.

Bledius semiferrugineus LeConte, J. L. – (2); TR; 3-4 May; hp.

Tribe Oxytelini

† *Anotylus rugosus* (Fabricius) – (3); DM, GF; 11 Apr-23 May; mt, pf.

Anotylus sp. – (4); AW, TR; 15 Apr, 30 Jun-24 Sep; bf, pf.

! *Apocellus sphaericollis* (Say) – (9); AW, Collingwood Picnic Area, GF, TR; bf, hp (along curb of parking lot), mt, pf.

† *Oxytelus laqueatus* (Marsham) – (1); TR; 31 Jul-17 Aug; mt.

! *Oxytelus pensylvanicus* Erichson – (1); GF; 1-15 Jul; mt. Fig. 5.

! † *Oxytelus sculptus* Gravenhorst – (1); TR; 18 Aug-4 Sep; mt.

Tribe Thinobiini

! † *Carpelimus bilineatus* Stephens – (7); DM, GF, TR; 15 Apr-14 Jun; hp, mt.

! *Carpelimus quadripunctatus* (Say) – (5); DM, GF, TR; 1 May-1 Aug; bl, hp, mt.

Carpelimus sp. – (1); DM; 15 Apr; hp.

! *Thinodromus arcifer* (LeConte, J. L.) – (1); DM; 19-28 Apr; mt. Fig. 6.

Subfamily Paederinae

Tribe Lathrobiini

Subtribe Lathrobiina

Lathrobium sp. – (1); TR; 1-20 May; mt.

! *Lobrathium collare* (Erichson) – (3); TR; 25 May, 9 Sep-21 Oct; hp, mt.

Pseudolathra sp. – (2); DM, LH; 10 Jun-8 Aug; hp, mt.

Tetartopeus sp. – (1); GF; 5 Jul; bl.

Subtribe Medonina

Achenomorphus corticinus (Gravenhorst) – (24); AW, DM, GF, TR; 15 Apr-30 Oct; bf, bt, mt.

! *Sunius confluentus* (Say) – (5); GF, LH; 13 Apr-15 May; hp (on tree trunk at night, under bark), mt.

! *Sciocharis carolinensis* Casey – (1); GF; 19-30 Jun; mt. Fig. 7.

! *Sciocharis exilis* (Erichson) – (1); AW; 15 Apr; bf. Fig. 8. This specimen documents a northern range extension from Alabama and Florida.

Subtribe Scopaeina

Scopaeus sp. – (8); DM, GF, TR; 15 Apr-18 May, 5 Aug-22 Oct; hp, mt.

Subtribe Stilicina

Eustilicus tristis (Melsheimer, F. E.) – (3); TR; 1 May-5 Jun; mt.

! *Rugilus angularis* (Erichson) – (1); GF; 15 Apr; bf.

Subtribe Stilicopsina

Stannoderus monstrosus (LeConte, J. L.) – (1); DM; 18 May; hp.

Tribe Paederini

Subtribe Cryptobiina

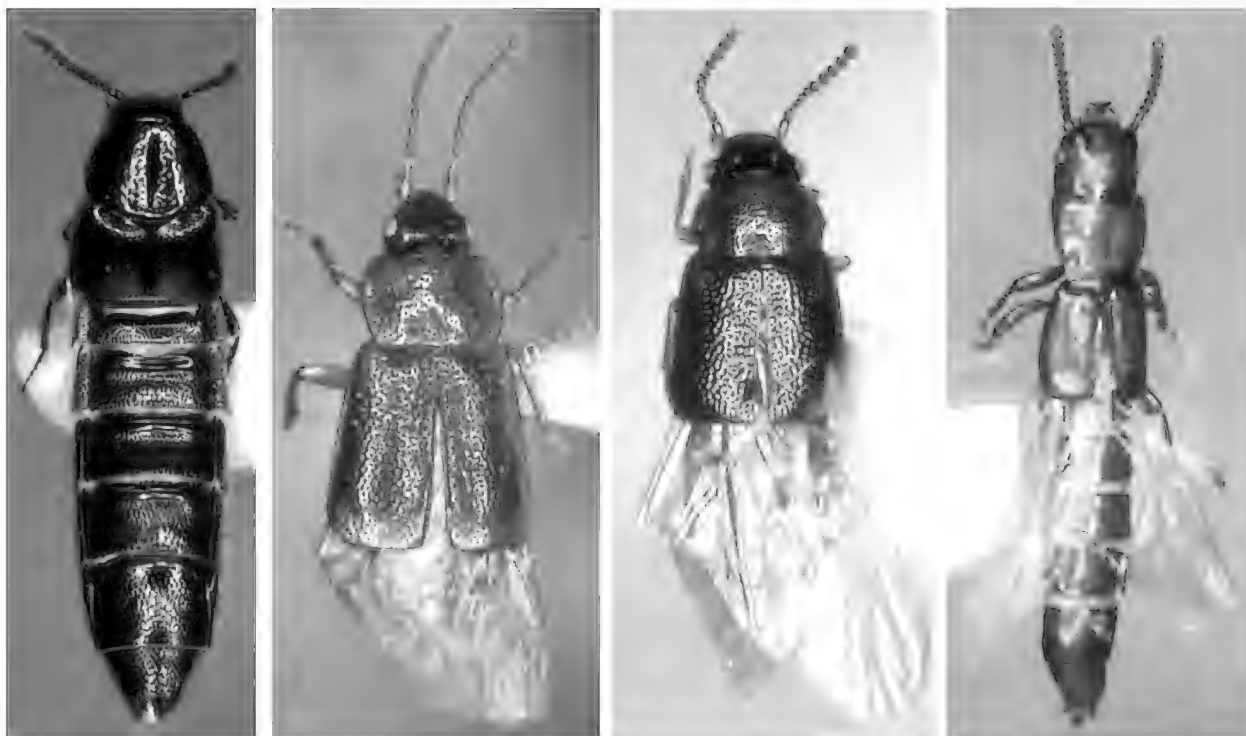
Homaeotarsus badius (Gravenhorst) – (13); GF, TR; 24 Apr-7 Sep; hp, mt.

Homaeotarsus bicolor (Gravenhorst) – (8); GF, TR; 20 May-17 Jul, 22 Oct-1 Dec; bt, hp, mt.

Homaeotarsus capito (Casey) – (2); AW, GF; 11-28 Apr; bf, lf.

Homaeotarsus carolinus (Erichson) – (1); GF; 11-28 Apr; pf.

! *Homaeotarsus cribratus* (LeConte, J. L.) – (2); TR; 19-21 Sep; pf.



Figs. 1-4 (left to right).



Figs. 5-8 (left to right).

! *Homaeotarsus pimerianus* (LeConte, J. L.) – (2); GF; 31 July; hp. Figs. 9-10. These specimens represent a significant eastern range extension from Indiana and Texas.

Subtribe Paederina

Paederus obliteratus (LeConte, J. L.) – (1); DM; 15 Apr; bf.

Tribe Pinophilini

Subtribe Pinophilina

Pinophilus latipes Gravenhorst – (2); GF; 23 Jun; bl.

Subfamily Piestinae

! *Siagonium americanum* (Melsheimer) – (1); LH; 3-16 Jun; lf. This first Virginia record fills a gap in a broad eastern distribution in North America.

Subfamily Proteininae

Tribe Proteinini

Proteinus sp. – (1♀); LH; 14-30 Jun; mt. This species is similar to the adventive species *P. atomarius* Erichson, reported from the District of Columbia, but the identity

could not be confirmed based on the single female.

Subfamily Pselaphinae

Supertribe Batrisitae

Tribe Batrisini

Subtribe Batrisina

Arthmius globicollis LeConte, J. L. – (5); DM, GF, TR; 14 Mar-15 Apr, 5 Aug-21 Oct; bf, hp, mt.

! *Batrisodes furcatus* (Brendel) – (2); TR; 23 May-5 Jun; mt. Fig. 11. This species has been documented north to New York and south to Alabama, so it was to be expected in Virginia.

Batrisodes lineaticollis (Aubé) – (1); GF; 21 May-18 Jun; mt.

! *Batrisodes scabriceps* (LeConte, J. L.) – (1); TR; 21 Jun-6 Jul; mt.

Batrisodes schaumii (Aubé) – (1); TR; 15-30 Oct; mt.

! *Batrisodes striatus* (LeConte, J. L.) – (1); GF; 11-28 Apr; pf.

Supertribe Euplectitae

Tribe Euplectini

Euplectus confluent LeConte, J. L. – (3); AW, FM; 15 Apr-14 May; bf.

Figure captions (opposite page)

Fig. 1. *Aleochara verna* Say – Dyke Marsh Wildlife Preserve, 13-20 June 1999, Malaise trap. Collected by E. Barrows. Determined by E. R. Hoebeke. Length 4.3 mm.

Fig. 2. *Arpedium* new species – Dyke Marsh Wildlife Preserve, 24 October-8 November 1999, Malaise trap. Collected by E. Barrows. Determined by M. K. Thayer. Length 4.1 mm.

Fig. 3. *Omalium* (sensu lato) *fractum* (Fauvel) – Turkey Run Park, 22 October-1 December 2009, Malaise trap. Collectors D. Smith & B. Steury. Determined by M. K. Thayer. Length 2.2 mm.

Fig. 4. *Clavilispinus rufescens* (LeConte, J. L.) – Little Hunting Creek, 13 April-15 May 2017, Malaise trap. Collectors C. Acosta, C. Davis, & B. W. Steury. Determined by R. M. Brattain. Length 3.5 mm.

Fig. 5. *Oxytelus pensylvanicus* Erichson – Great Falls Park, quarry site, 1-15 July 2008, Malaise trap. Collector D. Smith. Determined by R. M. Brattain. Length 2.4 mm.

Fig. 6. *Thinodromus arcifer* (LeConte, J. L.) – Dyke Marsh Wildlife Preserve, 19-28 April 1998, Malaise trap. Collector E. Barrows. Determined by R. M. Brattain. Length 2.5 mm.

Fig. 7. *Sciocharis carolinensis* Casey – Great Falls Park quarry site, 19-30 June 2009, Malaise trap. Collectors D. Smith & B. Steury. Determined by R. M. Brattain. Length 3.5 mm.

Fig. 8. *Sciocharis exilis* (Erichson) – Arlington Woods, Berlese funnel, 15 April 2013. Collectors M. S. Skvarla & J. R. Fisher. Determined R. M. Brattain. Length 2.5 mm.



Figs. 9-12 (left to right).

Figs. 9-10. *Homaeotarsus pimerianus* (LeConte, J. L.) – Great Falls Park at mouth of Difficult Run along Potomac River, hand collected by splashing water on silty gravel shoreline, 31 July 2017. Collector. B. W. Steury. Determined by R. M. Brattain & A. F. Newton. Length 9.8 mm. Left, anterior dorsal habitus, right, close up of head and pronotum.

Fig. 11. *Batrisodes furcatus* (Brendel) – Turkey Run Park, Malaise trap, 23 May-6 June 2008. Collector D. Smith. Determined by D. S. Chandler. Length 2.1 mm.

Fig. 12. *Neobisnius paederoides* (LeConte, J. L.) – Turkey Run Park, hand collected by splashing water on silt-caked leaf-litter along shore of Potomac River, 1 May 2017. Collector B. W. Steury. Determined by A. Brunke. Length 5.0 mm.

Tribe Trichonychini

Subtribe Panaphantina

Eutyphlus similis LeConte, J. L. – (1); GF; 14 Mar; hp.

Subtribe Trimiina

Melba parvula (LeConte, J. L.) – (2); DM; 18 May; hp.

Trimiomelba dubia (LeConte, J. L.) – (2); 16 May; FM; bf.

Tribe Trogastrini

Subtribe Rhexiina

Rhexius schmitti Brendel – (3); AW, FM, LH; 13 Apr-16 May; bf, mt.

Rhexius substriatus LeConte, J. L. – (2); DM. TR; 1-20 May; hp, mt.

Supertribe Goniaceritae

Tribe Brachyglutini

Subtribe Brachyglutina

Brachygluta abdominalis (Aubé) – (2); GF, TR; 18 Aug-30 Oct; mt.

Brachygluta luniger (LeConte, J. L.) – (7); DM, GF; 15 May-29 Jun; bf, mt.

Brachygluta ulkei (Brendel) – (12); DM; 15 Apr-18 May; bf.

Subtribe Pselaptina

Eutrichites zonatus (Brendel) – (1); DM; 15 Apr; bf.

Supertribe Pselaphitae

Tribe Ctenistini

Ctenisodes sp. – (1); DM; 15 Apr; bf.

Tribe Tmesiphorini

Tmesiphorus carinatus (Say) – (1); DM; 6 Jun; bf.

Tribe Tyrini

Subtribe Tyrina

! *Cedius spinosus* LeConte, J. L. – (1); LH; 19 May-2 Jun; mt

Subfamily Scaphidiinae

Tribe Cypariini

! *Cyparium concolor* (Fabricius) – (30); DM, GF, TR; 21 May-7 Sep; mt.

Tribe Scaphidiini

Scaphidium piceum Melsheimer, F. E. – (13); GF, TR; 21 May-30 Jul; hp, mt,

! *Scaphidium quadriguttatum* Say – (3); GF, LH, TR; 21 May-18 Jun, 11-28 Aug; hp (under log), mt.

Tribe Scaphisomatini

Baeocera sp. 1 – (2); TR; 1 May-30 Jun; mt.

Baeocera sp. 2 – (3); DM, GF; 16 Jul-7 Sep; mt.

Scaphisoma convexum Say – (3); GF, TR; 21 May-30 Jul; mt.

! *Scaphisoma terminatum* Melsheimer, F. E. – (1); GF; 31 Jul-17 Aug; mt.

Scaphisoma sp. 1 – (2); GF; 30 Jun-25 Aug; mt.

Scaphisoma sp. 2 – (1); TR; 3-17 Jul; mt.

Toxidium gammaroides LeConte, J. L. – (2); GF, TR; 16 Jul-17 Aug; mt.

Subfamily Scydmaeninae

Supertribe Cephenniitae

Tribe Cephenniini

Cephennodes cf. *virginicus* (Casey) – (6); AW, FM, GB; 15 Apr, 19-21 Oct; bf. Based on known ranges of closely allied species this taxon is most likely *C. virginicus* but it could be an undescribed species.

Supertribe Scydmaenitae

Tribe Chevrolatiini

Chevrolatia amoena LeConte, J. L. – (1); LH; 1-14 Jun; mt.

Tribe Glandulariini

Euconnus spp. – (8); AW, GF, Riverside Park, TR; 13 Apr-6 Jul, 19 Sep-21 Oct; bf, hp, mt.

! *Parascydmus* sp. – (1); LH; 19 Sep-10 Oct; mt. This genus has not been previously recorded from Virginia, Maryland, or the District of Columbia.

Subfamily Staphylininae

Tribe Diochini

Diochus schaumii Kraatz – (4); DM, GF; 15 Apr. bf, hp.

Tribe Staphylinini

Subtribe Acylophorina

! *Acylophorus agilis* Smetana – (4); DM; 19 Apr-9 Aug; mt.

! *Acylophorus caseyi* Leng – (2); GF; 1 May, 15 Aug-7 Sep; hp, mt. These first Virginia records represent a range extension for this mainly northern species which has been known from West Virginia and Pennsylvania to the north but has also been documented south to Alabama and Louisiana.

Acylophorus flavicollis Sachse – (1); GF; 1 Jun; hp (edge of vernal pool).

! *Hemiquedius infinitus* Brunke & Smetana – (1); GF; 5 Sep-21 Oct; mt.

Subtribe Anisolinina

Tympanophorus puncticollis (Erichson) – (1); GF; 19-30 Jun; mt.

Subtribe Erichsoniina

Erichsonius alumnus Frank, J. H. – (9); DM, GF, TR; 12 Apr-11 Jun; hp (swamp), mt.

Erichsonius brachycephalus Frank, J. H. – (4); GF, TR; 21 May-17 Jul; mt.

Subtribe Philonthina

Belonuchus rufipennis (Fabricius) – (19); DM, GF, TR; 14 Apr-26 Sep; hp, mt, pf.

Bisnius blandus (Gravenhorst) – (5); AW, GF, LH, TR; 15 Apr-18 May, 3 Jul-4 Sep; bf, mt, pf.

! *Bisnius pugetensis* (Hatch) – (1); TR; 21 Jul-4 Aug; mt. This first Virginia record represents a southern range extension from Pennsylvania.

Hesperus apicalis (Say) – (9); GF, LH, TR; 10 Apr-11 Aug; hp, lf, mt.

Hesperus baltimorensis (Gravenhorst) – (12); DM, GF, TR; 11 Apr-18 May, 1 Jul-26 Oct; hp, lf, mt.

! *Hesperus stehri* Moore – (1); TR; 28 Apr-12 May; pf. This specimen represents a northeastern range extension from North Carolina and Tennessee.

Laetulonthus laetulus (Say) – (2); GF; 11 Apr-19 May; lf.

Neobisnius jocosus (Horn) – (6); DM, GF, TR; 21 May-4 Sep; mt.

! *Neobisnius paederoides* (LeConte, J. L.) – (4); TR; 1-4 May; hp. Fig. 12.

Neobisnius sobrinus (Erichson) – (3); DM, TR; 28 Apr-10 May, hp, mt.

Neobisnius terminalis (LeConte, J. L.) – (1); TR; 4 May, hp.

Philonthus asper Horn – (51); AW, DM, GF, TR; 15 Apr-13 Oct; bf, hp, lf, mt, pf.

Philonthus caeruleipennis (Mannerheim) – (6); GF, TR; 21 May-21 Jul, 26 Sep; hp, mt.

† *Philonthus cognatus* Stephens – (2); TR; 1-21 Jul; mt.

† *Philonthus debilis* (Gravenhorst) – (1); TR; 19-21 Sep; pf.

Philonthus gracilior Casey – (2); GF, TR; 11-13 Jun; hp (swamp and splashing).

Philonthus lomatus Erichson – (7); DM, GF, TR; 28 May-21 Jul, 26 Sep; hp, mt.

! *Philonthus palliatus* (Gravenhorst) – (1); DM; 6-20 Jul; mt.

Philonthus quadricollis Horn – (2); TR; 1 May; hp.

Philonthus rufulus Horn – (21); DM, GF, LH, TR; 10 Apr-21 Jul, 15-30 Oct; bl, hp (under log and splashing), mt, pf.

Philonthus sericans (Gravenhorst) – (21); DM, GF, TR; 19 Apr-21 Oct; hp, mt.

Philonthus thoracicus (Gravenhorst) – (1); LH, 10 Jun; hp.

! *Philonthus umbrinoides* Smetana – (1); DM; 8-23 May; mt. This specimen documents a southern range extension from New York and West Virginia.

Philonthus validus Casey – (6); DM, GF, TR; 7 Jul-21 Oct; mt.

! † *Philonthus ventralis* (Gravenhorst) – (1); TR; 18 Mar-9 Apr; mt.

Subtribe Quediina

Quedius capucinus (Gravenhorst) – (3); GF; 19 Sep-21 Oct; mt.

† *Quedius mesomelinus* (Marsham) – (1); TR; 16-30 Jul; mt.

Quedius peregrinus (Gravenhorst) – (3); GF, TR; 21 May-21 Jul, 19 Sep-21 Oct; mt.

Subtribe Staphylinina

Platydracus cinnamopterus (Gravenhorst) – (8); AW, FM, GF, TR; 14 Apr-26 Jul; bf, hp (under bark), mt.

! *Platydracus exulans* (Erichson) – (1); GF; 10-30 Apr; mt.

Platydracus maculosus (Gravenhorst) – (36); AW, GF, LH, TR; Feb 5-12, 1 Jun-21 Oct; hp, lf, mt, pf.

Platydracus praetermissus Newton – (2); LH, GB; 15 Apr, 3-16 Jun; bf, pf.

! *Platydracus violaceus* (Gravenhorst) – (6); LH, GF, TR; 14 Apr-15 Jul; hp (under bark), mt, pf.

! *Platydracus viridanus* (Horn) – (1); TR; 21 Jul-4 Aug; mt.

Platydracus zonatus (Gravenhorst) – (6); AW, DM, FM, GB, TR; 15 Apr-25 May; bf, hp, pf.

! † *Tasgius winkleri* (Bernhauer) – (1); TR; 16 Sep; hp. This first Virginia record represents a southern range extension from Pennsylvania for this adventive species, which is slowly expanding its range in North America.

Tribe Xantholinini

Neohypnus emmesus (Gravenhorst) – (4); AW, DM, LH, GF; 14-29 Apr; bf, hp, lf, mt.

Neohypnus sp. – (2); AW, DM; 14 May, 9 Aug; bf, mt.

! *Oxybleptes kiteleyi* Smetana – (23); GF, TR; 16 Jul-21 Oct; mt. These first Virginia records represent a range extension for this rare species which was previously known only from New York and farther north, except for a record from North Carolina.

Subfamily Steninae

Stenus callosus Erichson – (10); DM, GF, TR; 1 May-4 Sep, 21 Nov-5 Dec; hp, mt.

Stenus colon Say – (3); GF, TR; 24 Apr-1 Jun; hp.

Stenus colonus Erichson – (6); DM, TR; 4 May-14 Jun; hp, mt.

! *Stenus croceatus* (Casey) – (2); DM; 18-30 May; hp, mt.

Stenus egenus Erichson – (1); TR; 20 Apr; hp

Stenus femoratus Say – (4); DM, TR; 1 May, 26 Sep-11 Oct; hp, mt.

Stenus sp. – (1); DM; 18 May; hp.

Subfamily Tachyporinae**Tribe Mycetoporini**

! † *Bolitobius cingulatus* Mannerheim – (1); GF; 18 Aug-4 Sep; mt. This Virginia specimen represents a range extension for this adventive species which is also known from New Jersey and farther north, and from Alabama and Florida to the south.

! *Bryophacis smetanai* Campbell – (1); TR; 19-30 Jun; mt. This record represents a southern range extension from Pennsylvania.

Bryoporus rufescens LeConte, J. L. – (26); DM, GF, TR; 1 May-21 Oct; mt.

Bryoporus testaceus LeConte, J. L. – (1); GF; 20 May; hp.

! *Lordithon appalachianus* Campbell – (14); GF, TR; 1 May-1 Dec; mt.

Lordithon cinctus (Gravenhorst) – (11); AW, GF, LH, TR; 15 Apr-21 Oct; bf, mt.

Lordithon facilis (Casey) – (8); AW, GF, TR; 14 May-15 Jul, 5 Sep-21 Oct; bf, mt.

Lordithon kelleyi (Malkin) – (8); DM, GF, TR; 1 May-21 Oct; mt.

Lordithon niger (Gravenhorst) – (1); TR; 1-15 Jul; mt.

Lordithon notabilis Campbell – (3); GF, TR; 19 Jun-21 Oct; mt.

Lordithon quaesitor (Horn) – (2); TR; 19-30 Jul; mt.

Mycetoporus americanus Erichson – (4); GF, TR; 10-30 Apr, 7 Jul-4 Sep; mt.

Mycetoporus lucidulus LeConte, J. L. – (3); GF, TR; 10-30 Apr, 16 Jul-17 Aug; mt.

Tribe Tachyporini

Coproporus laevis LeConte, J. L. – (11); GF; 25 Jun, 19 Sep-21 Oct; bl, mt.

Coproporus ventriculus (Say) – (11); LH, GF, TR; 12 Apr-7 Sep; bl, hp, lf, mt.

! *Nitidotachinus scrutator* (Gemminger & Harold) – (2); TR; 19 Jun-15 Jul; mt.

Sepedophilus basalis (Erichson) – (2); FM, GF; 16 May-29 Jun; hp, mt.

Sepedophilus cinctulus (Erichson) – (1); TR; 5-25 Aug; mt.

Sepedophilus crassus (Gravenhorst) – (33); Fort Hunt Park, GF, TR; 15 Jun-1 Dec; hp, mt.

Sepedophilus frosti Campbell – (4); DM, GF, TR; 24 Jun-30 Jul, 22 Oct-1 Dec; mt.

† *Sepedophilus littoreus* (Linnaeus) – (13); AW, DM, GF, TR; 10 Apr-4 Sep; mt.

! *Sepedophilus occultus* (Casey) – (1); GF; 15-29 Jun; mt.

Sepedophilus opicus (Say) – (9); AW, FM, GF, TR; 15 Apr-20 Jul; bf, hp, mt.

Sepedophilus scriptus (Horn) – (1); GF; 21 May-18 Jun; mt.

Sepedophilus velocipes (Casey) – (2); TR; 2 Jul-4 Sep; mt.

Sepedophilus versicolor (Casey) – (13); AW, LH, GF, TR; 11 Apr-21 Oct; bf, lf, mt, pf.

Tachinus axillaris Erichson – (3); GF, TR; 27 Apr-19 May, 7-30 Jul; mt, pf.

Tachinus canadensis Horn – (3); GF, TR; 13 Oct-1 Dec; mt, pf.

Tachinus fimbriatus Gravenhorst – (27); DM, GF, TR; 21 May-7 Jul, 19 Sep-5 Dec; bl, mt, pf.

Tachinus fumipennis (Say) – (6); GF, TR; 7 Jun-30 Jul, 5 Sep-21 Oct; mt, pf.

Tachinus minimus Campbell – (1); TR; 19-30 Jun; mt.

Tachyporus jocosus Say – (1); AW; 14 May; bf.

! *Tachyporus lecontei* Campbell – (1); GB; 17 Jun; hp. This specimen represents a southern range extension from Pennsylvania.

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APPENDIX

**Preliminary Checklist of Rove Beetles
(Coleoptera: Staphylinidae) of Virginia,
Maryland, and the District of Columbia**

Alfred F. Newton (compiler)

The following checklist includes all species or subspecies of Staphylinidae that are currently known to occur in Virginia, Maryland and the District of Columbia, including those from Virginia newly reported in this work. The list contains 792 taxa including 558 from Virginia, 343 from Maryland, and 494 from the District of Columbia. Sixty species are believed to be adventive in North America, originating from the Palaearctic region and in most cases probably from Europe, with the exception of *Anotylus insignitus* from the Neotropical region. Another fifteen species are Holarctic (or even more broadly distributed). One hundred and fourteen species are first records for Virginia, eight are new to Maryland and four are new to the District of Columbia. The Checklist is extracted from a world catalog database compiled and maintained by AFN, which is now available online in simplified form via the Catalogue of Life (Newton 2019). The inclusion of these states and district in the distribution of these taxa is based, with rare exceptions, on published sources, which include the original descriptions of each name (all were consulted), and secondary sources including the catalogs of Horn (1868), Ulke (1902), Leng (1920), Blackwelder (1973a, 1973b), Moore & Legner (1975), Chandler (1997), Herman (2001), Gusarov (2003) and Löbl (2018), recent regional reviews including Downie & Arnett (1996), Newton et al. (2000), O'Keefe (2000), Brown (2008), Brunke et al. (2011), and Klimaszewski et al. (2013, 2018), and generic revisions, monographs, notes or similar sources concerning many taxa, most of which are listed in the individual generic treatments for Staphylinidae in American Beetles (Newton et al. 2000, O'Keefe, 2000). A few previously unreported state records (marked below as new) are included based on still-unpublished revisionary work or identifications by AFN or others as indicated, and additional new records (also marked as such) are included based on the above GWMP study. In spite of efforts to make this list as complete and accurate as possible, there are undoubtedly additional published records that were overlooked, and some taxa included in the list may have been erroneously reported from these areas. Based on what is known about the overall distribution patterns of staphylinids in the eastern United States, many additional species can be expected to actually occur in these states or district but, to our knowledge, have not yet been found or reported

from there; the actual staphylinid diversity of this area is no doubt much larger than shown in this list. Thus, we consider this checklist a preliminary one, in need of further review and documentation. The format of the checklist is adapted from Bousquet et al. (2013).

Further information about each species or subspecies listed here, including the original generic combination (if different from the current one), a reference to the original description, synonyms (if any), and approximate overall distribution, can be found in the Catalogue of Life (Newton, 2019).

Abbreviations**Areas:**

DC	District of Columbia
MD	Maryland (state)
VA	Virginia (state)

Identifiers (for new state or district records):

AFN	Alfred F. Newton, FMNH, Chicago, Illinois
AJB	Adam J. Brunke, Canadian National Collection of Insects, Arachnids and Nematodes, Ottawa, Canada
AVE	Arthur V. Evans, Richmond, VA (also personal collection)
CWH	Curt W. Harden, VMNH, Martinsville, VA (also personal collection)
DSC	Donald S. Chandler, University of New Hampshire, Durham, New Hampshire (also personal collection)
ERH	E. Richard Hoebeke, Georgia Museum of Natural History, Athens, Georgia
MKT	Margaret K. Thayer, FMNH, Chicago, Illinois
MLF	Michael L. Ferro, Clemson University, Clemson, South Carolina

Collections:

AMNH	American Museum of Natural History, New York, NY
BMNH	The Natural History Museum, London, Great Britain
CNCI	Canadian National Collection of Insects, Ottawa, Ontario, Canada
CUAC	Clemson University, Clemson, South Carolina
DENH	Department of Biological Sciences, University of New Hampshire, Durham, New Hampshire

DFOC	USFS Durham Field Office Forest Insect Collection, Durham, New Hampshire
EGRC	E. G. Riley collection, College Station, Texas
EIUC	Eastern Illinois University, Charleston, Illinois (as of 12/11/2019 part of FMNH)
EJFC	E. J. Ford collection (present location unknown)
FMNH	Field Museum of Natural History, Chicago, Illinois
GWMP	George Washington Memorial Parkway collection (property of U.S. National Park Service), McLean, VA
MCZC	Museum of Comparative Zoology, Harvard University, Cambridge, Massachusetts
UMSP	University of Minnesota, St. Paul, Minnesota
USNM	National Museum of Natural History, Washington, DC
VMNH	Virginia Museum of Natural History, Martinsville, VA
VPIC	Virginia Polytechnic Institute and State University, Blacksburg, VA

Species are listed in alphabetical sequence by higher taxon, genus and species. Subgenera are indicated only when widely used in North America or when sometimes treated as genera, but do not affect the species sequence. Species that are believed to be adventive (“introduced”) in North America (from the Palearctic region, with the exception of the Neotropical *Anotylus insignitus*) are indicated by a dagger (†). Those that are considered to be Holarctic (or even more broadly distributed) are indicated by an asterisk (*). New state or district records are boldfaced and marked with an exclamation point, and the source(s) of such records indicated in the “Source” column using the above abbreviations.

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Subfamily	Supertribe	Tribe	Subtribe	Name	DC	MD	VA	Source
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Coprochara) bimaculata</i> Gravenhorst, 1802	DC	MD	VA	
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Xenochara) castaneimarmotae</i> Klimaszewski, Webster & Brunke, 2017	-	-	VA!	VA-CWH in VMNH
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Xenochara) castaneipennis</i> Mannerheim, 1843	-	MD	-	
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Aleochara) curtula</i> (Goeze, 1777) [†]	DC	-	-	
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Xenochara) fumata</i> Gravenhorst, 1802 [†]	-	MD	VA	
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Aleochara) gracilicornis</i> Bernhauer, 1901	-	MD	VA!	VA-CWH in CWH
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Aleochara) lata</i> Gravenhorst, 1802 [†]	DC	MD	VA	
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Emplenota) litoralis</i> (Mäklin, 1853)	-	-	VA	
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Echochara) lucifuga</i> (Casey, 1893)	-	MD	VA	
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Aleochara) lustrica</i> Say, 1832	DC	MD	VA	
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Echochara) ocularis</i> Klimaszewski, 1984	-	-	VA!	VA-ERH in GWMP
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Calochara) rubripes</i> Blatchley, 1910	-	-	VA!	VA-ERH in GWMP
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Xenochara) sculptiventris</i> (Casey, 1893)	DC	-	-	
Aleocharinae		Aleocharini	Aleocharina	<i>Aleochara (Coprochara) verna</i> Say, 1833*	DC	-	VA!	VA-ERH in GWMP
Aleocharinae		Athetini		<i>Apalonia seticornis</i> Casey, 1906	-	-	VA!	VA-ERH in GWMP
Aleocharinae		Athetini		<i>Goniusa caseyi</i> Gusarov, 2003	DC	-	-	
Aleocharinae		Athetini		<i>Halobrecthina opaciceps</i> (Bernhauer, 1909)	-	-	VA	
Aleocharinae		Athetini		<i>Halobrecthina sibylla</i> (Casey, 1910)	-	-	VA	
Aleocharinae		Athetini		<i>Meroneira venustula</i> (Erichson, 1839)	DC	-	VA!	VA-CWH in CWH
Aleocharinae		Athetini	Athetina	<i>Acrotoma curata</i> (Casey, 1910)	-	-	VA	
Aleocharinae		Athetini	Athetina	<i>Acrotoma recondita</i> (Erichson, 1839)	DC	-	-	
Aleocharinae		Athetini	Athetina	<i>Amischa analis</i> (Gravenhorst, 1802) [†]	DC	-	-	
Aleocharinae		Athetini	Athetina	<i>Atheta (Atheta) aemula</i> (Erichson, 1839)	DC	-	-	
Aleocharinae		Athetini	Athetina	<i>Atheta (Pseudota) annexa</i> Casey, 1910	-	-	VA	
Aleocharinae		Athetini	Athetina	<i>Atheta (Atheta) castanoptera</i> (Mannerheim, 1830) [†]	-	MD	-	
Aleocharinae		Athetini	Athetina	<i>Atheta (Dimetrota) modesta</i> (F.E. Melsheimer, 1844)	DC	-	-	

Aleocharinae	Athetini	Athetina	<i>Atheta (Microdota) pennsylvanica</i> Bernhauer, 1907	-	-	VA
Aleocharinae	Athetini	Athetina	<i>Atheta (Noverota) personata</i> (Casey, 1910)	-	-	VA
Aleocharinae	Athetini	Athetina	<i>Atheta restricta</i> Casey, 1911	-	-	VA
Aleocharinae	Athetini	Athetina	<i>Atheta reticula</i> Casey, 1910	-	-	VA
Aleocharinae	Athetini	Athetina	<i>Atheta texana</i> Casey, 1910	-	-	VA
Aleocharinae	Athetini	Athetina	<i>Atheta (Dimetrota) trogliphila</i> Klimaszewski & Peck, 1986	-	-	VA
Aleocharinae	Athetini	Athetina	<i>Atheta (Alaobia) ventricosa</i> Bernhauer, 1907	DC	-	-
Aleocharinae	Athetini	Athetina	<i>Euromota lucida</i> Casey, 1906	-	-	VA
Aleocharinae	Athetini	Athetina	<i>Lypoglossa angularis obtusa</i> (J.L. LeConte, 1866)	DC	-	-
Aleocharinae	Athetini	Athetina	<i>Microtheta caudex</i> (Casey, 1910)	-	-	VA
Aleocharinae	Athetini	Athetina	<i>Mocyta luteola</i> (Erichson, 1839)	DC	-	-
Aleocharinae	Athetini	Athetina	<i>Nehemitropia lividipennis</i> (Mannerheim, 1830)†	DC	-	-
Aleocharinae	Athetini	Athetina	<i>Stethusa dichroa</i> (Gravenhorst, 1802)	DC	MD	VA
Aleocharinae	Athetini	Athetina	<i>Trichiusa compacta</i> Casey, 1893	DC	-	-
Aleocharinae	Athetini	Athetina	<i>Trichiusa hirsuta</i> Casey, 1906	-	MD	VA
Aleocharinae	Athetini	Athetina	<i>Trichiusa polita</i> Casey, 1906	DC	-	-
Aleocharinae	Athetini	Athetina	<i>Trichiusa robustula</i> Casey, 1893	-	MD	-
Aleocharinae	Athetini	Athetina	<i>Trichiusa varicolor</i> Casey, 1906	-	-	VA
Aleocharinae	Athetini	Athetina	<i>Trichiusa virginica</i> Casey, 1906	-	-	VA
Aleocharinae	Falagriini	Falagriini	<i>Aleodorus bilobatus</i> Say, 1833	DC	MD	VA
Aleocharinae	Falagriini	Falagriini	<i>Aleodorus partitus</i> (J.L. LeConte, 1866)	-	-	VA
Aleocharinae	Falagriini	Falagriini	<i>Aleodorus scutellaris</i> (J.L. LeConte, 1866)	-	MD	-
Aleocharinae	Falagriini	Falagriini	<i>Borboropora quadriceps</i> (J.L. LeConte, 1866)	DC	MD	VA
Aleocharinae	Falagriini	Falagriini	<i>Cordalia obscura</i> (Gravenhorst, 1802)†	-	MD	-
Aleocharinae	Falagriini	Falagriini	<i>Falagria caesa</i> Erichson, 1837†	-	MD	VA
Aleocharinae	Falagriini	Falagriini	<i>Falagria dissecta</i> Erichson, 1839	DC	MD	VA
Aleocharinae	Falagriini	Falagriini	<i>Lissagria laticeps</i> (Notman, 1920)	-	-	VA
Aleocharinae	Falagriini	Falagriini	<i>Myrmecoccephalus cingulatus</i> (J.L. LeConte, 1866)	DC	MD	VA
Aleocharinae	Falagriini	Falagriini	<i>Myrmecoccephalus concinnus</i> (Erichson, 1839)*	-	MD	VA
Aleocharinae	Falagriini	Falagriini	<i>Myrmecoccephalus gracilis</i> (Casey, 1906)	-	-	VA
Aleocharinae	Geostibini	Geostibini	<i>Aloconota insecta</i> (C.G. Thomson, 1856)*	-	-	VA

Aleocharinae	Geostibini		<i>Aloconota sulcifrons</i> (Stephens, 1832)*	-	-	VA
Aleocharinae	Geostibini		<i>Eurota dentata</i> (Bernhauer, 1906)	DC	MD	VA
Aleocharinae	Geostibini		<i>Geostiba appalachigena</i> Gusarov, 2002	-	MD	VA
Aleocharinae	Gymnusiini		<i>Adlinopsis cuspidata</i> Klimaszewski, 1982	-	MD	-
Aleocharinae	Gymnusiini		<i>Adlinopsis myllaenoides</i> (Kraatz, 1857)	DC	-	-
Aleocharinae	Gymnusiini		<i>Deinopsis americanus</i> Kraatz, 1857	DC	-	-
Aleocharinae	Gymnusiini		<i>Deinopsis rapida</i> Casey, 1911	-	MD	VA
Aleocharinae	Gymnusiini		<i>Deinopsis rhadina</i> Klimaszewski, 1979	-	-	VA
Aleocharinae	Gymnusiini		<i>Deinopsis virginiana</i> Klimaszewski, 1979	-	-	VA
Aleocharinae	Gymnusiini		<i>Gymnusa grandiceps</i> Casey, 1915	-	MD	-
Aleocharinae	Homalotini	Bolitocharina	<i>Leptusa (Eucryptusa) brevicollis</i> Casey, 1893	-	-	VA
Aleocharinae	Homalotini	Bolitocharina	<i>Leptusa (Ulittusa) cribratula</i> (Casey, 1906)	-	-	VA
Aleocharinae	Homalotini	Bolitocharina	<i>Leptusa (Adoxopisalia) elegans</i> Blatchley, 1910	-	-	VA
Aleocharinae	Homalotini	Bolitocharina	<i>Leptusa (Adoxopisalia) opaca</i> Casey, 1893	DC	MD	-
Aleocharinae	Homalotini	Bolitocharina	<i>Pleurotobia trimaculata</i> (Erichson, 1839)	DC	-	-
Aleocharinae	Homalotini	Gyrophaenina	<i>Eumicrota corrugata</i> (Erichson, 1839)	DC	-	VA
Aleocharinae	Homalotini	Gyrophaenina	<i>Eumicrota socia</i> (Erichson, 1839)	DC	MD	VA
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena affinis</i> Mannerheim, 1830†	DC	-	VA
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena blackwelderi</i> Seevers, 1951	DC	-	VA
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena coniventris</i> Casey, 1906	DC	-	-
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena flavicornis</i> F.E. Melsheimer, 1844	DC	MD	VA
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena fuscicollis</i> Casey, 1906	DC	-	-
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena gilvicollis</i> Casey, 1906	DC	-	VA
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena hubbardi</i> Seevers, 1951	DC	-	-
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena laetula</i> Casey, 1906	DC	-	VA
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena lobata</i> Casey, 1906	DC	-	-
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena nanoides</i> Seevers, 1951	DC	-	VA
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena simulans</i> Seevers, 1951	-	MD	-
Aleocharinae	Homalotini	Gyrophaenina	<i>Gyrophaena vitrina</i> Casey, 1906	-	-	VA!
Aleocharinae	Homalotini	Gyrophaenina	<i>Phanerota dissimilis</i> (Erichson, 1839)	-	-	VA-CWH in CWH
Aleocharinae	Homalotini	Gyrophaenina		-	-	VA

Aleocharinae	Homalotini	Gyrophaenina	<i>Phanerota fasciata</i> (Say, 1832)	DC	MD	VA
Aleocharinae	Homalotini	Homalotina	<i>Homalota plana</i> (Gyllenhal, 1810) [†]	DC	-	-
Aleocharinae	Homalotini	Homalotina	<i>Thecturota capito</i> Casey, 1893	-	-	VA
Aleocharinae	Hoplandriini	Hoplandriina	<i>Hoplandria (Lophomnucter) klimaszewskii</i> Génier, 1989	DC	MD	VA
Aleocharinae	Hoplandriini	Hoplandriina	<i>Hoplandria (Lophomnucter) laevicollis</i> (Notman, 1920)	DC	-	VA
Aleocharinae	Hoplandriini	Hoplandriina	<i>Hoplandria (Arrhenandria) laeviventris</i> Casey, 1910	DC	MD	VA
Aleocharinae	Hoplandriini	Hoplandriina	<i>Hoplandria (Hoplandria) lateralis</i> (F.E. Melsheimer, 1844)	DC	MD	VA
Aleocharinae	Hoplandriini	Platandriina	<i>Microtia silacea</i> (Erichson, 1839)	-	-	VA
Aleocharinae	Hoplandriini	Platandriina	<i>Platandria carolinae</i> Casey, 1910	DC	-	VA
Aleocharinae	Hypocyphitini		<i>Oligota pumilio</i> Kiesenwetter, 1858 [†]	DC	-	-
Aleocharinae	Lomechusini	Lomechusina	<i>Xenodusa cava</i> (J.L. LeConte, 1863)	DC	MD	VA
Aleocharinae	Lomechusini	Lomechusina	<i>Xenodusa reflexa</i> (Walker, 1866)	-	MD	-
Aleocharinae	Lomechusini	Myrmedoniina	<i>Drusilla ashei</i> Eldredge, 2012	-	-	VA! VA-CWH in CWH
Aleocharinae	Lomechusini	Myrmedoniina	<i>Ecitoxenidia brevicornis</i> SeEVERS, 1959	-	-	VA
Aleocharinae	Lomechusini	Myrmedoniina	<i>Pella caliginosa</i> (Casey,)	DC	-	-
Aleocharinae	Lomechusini	Myrmedoniina	<i>Pella loricata</i> (Casey, 1893)	DC	-	-
Aleocharinae	Lomechusini	Myrmedoniina	<i>Zyras (Zyras) planifer</i> (Casey, 1893)	DC	-	-
Aleocharinae	Lomechusini	Myrmedoniina	<i>Zyras (Zyras) rudis</i> (J.L. LeConte, 1866)	DC	-	-
Aleocharinae	Mesoporini		<i>Anacryptus testaceus</i> (J.L. LeConte, 1863)	DC	-	-
Aleocharinae	Myllaenini		<i>Myllaena cuneata</i> Notman, 1920	-	MD	VA
Aleocharinae	Myllaenini		<i>Myllaena infusca</i> Kraatz, 1853*	DC	-	-
Aleocharinae	Myllaenini		<i>Myllaena kaskaskia</i> Klimaszewski, 1982	-	-	VA
Aleocharinae	Myllaenini		<i>Myllaena potawatomi</i> Klimaszewski, 1982	-	-	VA
Aleocharinae	Myllaenini		<i>Myllaena procliva</i> Casey, 1911	-	MD	VA
Aleocharinae	Oxypodini	Dinardina	<i>Decusa expansa</i> (J.L. LeConte, 1866)	-	MD	VA! VA-CWH in CWH
Aleocharinae	Oxypodini	Dinardina	<i>Euthorax debilis</i> (Wasmann, 1894)	DC	-	-
Aleocharinae	Oxypodini	Microglottina	<i>Cratarea suturalis</i> (Mannerheim, 1830) [†]	DC	-	VA
Aleocharinae	Oxypodini	Microglottina	<i>Haploglossa barberi</i> (Fenyès, 1921)	-	-	VA
Aleocharinae	Oxypodini	Oxypodina	<i>Dexiogyia angustiventris</i> (Casey, 1893)	DC	-	-
Aleocharinae	Oxypodini	Oxypodina	<i>Ocyusa asperula</i> Casey, 1893	DC	-	-

Aleocharinae	Oxypodini	Oxypodina	<i>Oxypoda mimetica</i> Casey, 1906	-	-	VA
Aleocharinae	Oxypodini	Oxypodina	<i>Oxypoda nigriceps</i> Casey, 1893	-	-	VA
Aleocharinae	Oxypodini	Oxypodina	<i>Oxypoda obliqua</i> Casey, 1906	-	-	VA
Aleocharinae	Oxypodini	Oxypodina	<i>Oxypoda sagulata</i> Erichson, 1839	DC	-	-
Aleocharinae	Oxypodini	Phloeoporina	<i>Phloeopora corticalis</i> (Gravenhorst, 1802)*	DC	-	-
Aleocharinae	Oxypodini	Phloeoporina	<i>Phloeopora sublaevis</i> Casey, 1906	DC	-	-
Aleocharinae	Philotermitini		<i>Philoterme cubitopilis</i> SeEVERS, 1957	DC	-	VA! VA-AVE in AVE
Aleocharinae	Philotermitini		<i>Philoterme fuchsii</i> Kraatz, 1857	-	-	VA
Aleocharinae	Philotermitini		<i>Philoterme pensylvanicus</i> Kraatz, 1857	DC	-	-
Aleocharinae	Philotermitini		<i>Philoterme pilosus</i> Kraatz, 1857	DC	-	-
Aleocharinae	Placusini		<i>Euvira micmac</i> Klimaszewski & Majka, 2007	-	-	VA! VA-CWH in CWH
Aleocharinae	Placusini		<i>Placusa despecta</i> Erichson, 1839	DC	-	-
Aleocharinae	Placusini		<i>Placusa tacomae</i> Casey, 1893	-	-	VA! VA-CWH in CWH
Aleocharinae	Tachyusini		<i>Gnypeta baltifera</i> (J.L. LeConte, 1863)	DC	-	-
Aleocharinae	Tachyusini		<i>Gnypeta nigrella</i> (J.L. LeConte, 1863)	DC	MD	-
Aleocharinae	Tachyusini		<i>Tachyusa americana</i> Casey, 1906	DC	MD	-
Aleocharinae	Tachyusini		<i>Tachyusa cavicollis</i> J.L. LeConte, 1863	-	MD	VA
Aleocharinae	Tachyusini		<i>Tachyusa faceta</i> Casey, 1885	DC	-	-
Aleocharinae	Tachyusini		<i>Tachyusa gracillima</i> J.L. LeConte, 1863	DC	MD	VA
Aleocharinae	Tachyusini		<i>Tachyusa obsoleta</i> Casey, 1906	DC	-	-
Aleocharinae	Taxicerini		<i>Halobrecta flavipes</i> C.G. Thomson, 1861†	-	-	VA
Dasycterinae			<i>Dasycerus carolinensis</i> Horn, 1882	-	-	VA
Dasycterinae			<i>Dasycerus concolor</i> Löbl & Calame, 1996	-	-	VA
Euaesthetinae	Euaesthetini		<i>Edaphus americanus</i> Puthz, 1974	-	-	VA
Euaesthetinae	Euaesthetini		<i>Edaphus nitidus</i> Motschulsky, 1857	DC	-	VA
Euaesthetinae	Euaesthetini		<i>Euaesthetus americanus</i> Erichson, 1840	DC	-	VA
Euaesthetinae	Euaesthetini		<i>Euaesthetus iripennis</i> Casey, 1884	DC	MD	VA
Euaesthetinae	Euaesthetini		<i>Euaesthetus mundulus</i> Casey, 1884	DC	-	VA
Euaesthetinae	Euaesthetini		<i>Euaesthetus punctatus</i> Casey, 1884	-	-	VA
Euaesthetinae	Euaesthetini		<i>Euaesthetus similis</i> Casey, 1884	-	MD	-

Euaesthetinae	Stictocraniini	<i>Stictocranius puncticeps</i> J.L. LeConte, 1866	DC	MD	VA
Megalopsidiinae		<i>Megalopinus caelatus</i> (Gravenhorst, 1802)	DC	-	VA
Megalopsidiinae		<i>Megalopinus rufipes</i> (Motschulsky, 1857)	-	MD	VA
Micropeplinae		<i>Micropeplus browni</i> Campbell, 1968	-	MD	-
Micropeplinae		<i>Micropeplus cribratus</i> J.L. LeConte, 1863	DC	-	-
Omalinae	Anthophagini	<i>Acidota crenata</i> (Fabricius, 1792)*	-	-	VA
Omalinae	Anthophagini	<i>Acidota subcarinata</i> Erichson, 1840	DC	MD	VA
Omalinae	Anthophagini	<i>Anthobium sordidum</i> (Erichson, 1840)	-	-	VA
Omalinae	Anthophagini	<i>Arpedium schwarzi</i> Fauvel, 1878	DC	-	VA
Omalinae	Anthophagini	<i>Brathinus nitidus</i> J.L. LeConte, 1852	-	MD	VA
Omalinae	Anthophagini	<i>Geodromicus brunneus</i> (Say, 1823)	DC	MD	VA
Omalinae	Anthophagini	<i>Geodromicus strictus</i> Fauvel, 1889	-	-	VA! VA-MKT in FMNH
Omalinae	Anthophagini	<i>Lesteva cribratula</i> (Casey, 1893)	-	-	VA
Omalinae	Anthophagini	<i>Lesteva pallipes</i> J.L. LeConte, 1863	DC	MD	VA
Omalinae	Anthophagini	<i>Olophrum consimile</i> (Gyllenhal, 1810)*	-	-	VA
Omalinae	Anthophagini	<i>Olophrum obtectum</i> Erichson, 1840	DC	MD	VA
Omalinae	Anthophagini	<i>Trigonodenus striatus</i> J.L. LeConte, 1863	-	-	VA
Omalinae	Coryphiini	<i>Ephelinus guttatus</i> (J.L. LeConte, 1863)	DC	-	VA
Omalinae	Coryphiini	<i>Ephelinus notatus</i> (J.L. LeConte, 1863)	DC	MD	VA
Omalinae	Coryphiini	<i>Eudectus crassicornis</i> J.L. LeConte, 1884	DC!	-	- DC-MKT in USFS
Omalinae	Eusphalerini	<i>Eusphalerum carolinensis</i> Zanetti, 2014	-	-	VA
Omalinae	Eusphalerini	<i>Eusphalerum convexum</i> (Fauvel, 1878)	DC	-	VA
Omalinae	Eusphalerini	<i>Eusphalerum horni</i> (Fauvel, 1878)	-	-	VA
Omalinae	Omalini	<i>Acrolocha diffusa</i> (Fauvel, 1878)	DC	-	-
Omalinae	Omalini	<i>Carcinocephalus flavidus</i> (Hamilton, 1895)	DC	MD	VA! VA-MKT in VMNH
Omalinae	Omalini	<i>Hapalareoa hamata</i> (Fauvel, 1878)	DC	-	VA! VA-MKT in USNM
Omalinae	Omalini	<i>Omalium cribrum</i> Fauvel, 1878	DC	-	VA! VA-MKT in FMNH
Omalinae	Omalini	<i>Omalium quadripenne</i> Casey, 1893	-	-	VA

Omalinae	Omalini	<i>Omalium repandum</i> Erichson, 1840	DC	-	VA!	VA-GWMP, MKT in USNM
Omalinae	Omalini	<i>Omalium rivulare</i> (Paykull, 1789) [†]	-	-	VA	
Omalinae	Omalini	<i>Omalium</i> (sensu lato) <i>fractum</i> Fauvel, 1878	DC	-	VA!	VA-MKT in GWMP, VMNH
Omalinae	Omalini	<i>Phloeonormus laescollis</i> (Mäklin, 1852)	DC	-	VA!	VA-MKT in USNM
Omalinae	Omalini	<i>Phyllodrepa humerosa</i> (Fauvel, 1878)	DC	MD!	VA!	MD-MKT in UMSP; VA-GWMP, MKT in USNM
Omalinae	Omalini	<i>Phyllodrepa punctiventris</i> (Fauvel, 1878)	-	-	VA	
Omalinae	Omalini	<i>Xylodromus concinnus</i> (Marsham, 1802) [†]	-	MD	VA!	VA-GWMP
Omalinae	Omalini	<i>Xylodromus suteri</i> Thayer, 2003	-	MD	VA	
Osoiriinae	Eleusinini	<i>Eleusis pallida</i> (J.L. LeConte, 1863)	DC	-	VA!	VA-GWMP
Osoiriinae	Eleusinini	<i>Renardia nigrella</i> (J.L. LeConte, 1863)	DC	-	-	
Osoiriinae	Osoirini	<i>Holotrochus brachypterus</i> Fauvel, 1905	-	-	VA	
Osoiriinae	Osoirini	<i>Molosoma latipes</i> (Gravenhorst, 1806)	DC	MD	VA	
Osoiriinae	Thoracophorini	<i>Clavilispinus exiguus</i> (Erichson, 1840)	DC	-	-	
Osoiriinae	Thoracophorini	<i>Clavilispinus rufescens</i> (J.L. LeConte, 1863)	-	-	VA!	VA-GWMP
Osoiriinae	Thoracophorini	<i>Prolibia laevicauda</i> (J.L. LeConte, 1866)	DC	-	VA	
Osoiriinae	Thoracophorini	<i>Thoracophorus costalis</i> (Erichson, 1840)	DC	MD	VA	
Oxyporinae	Thoracophorini	<i>Oxyporus femoralis femoralis</i> Gravenhorst, 1802	DC	MD	VA	
Oxyporinae		<i>Oxyporus lepidus</i> J.L. LeConte, 1877	DC	MD	VA	
Oxyporinae		<i>Oxyporus major</i> Gravenhorst, 1806	DC	MD	VA	
Oxyporinae		<i>Oxyporus rufipennis</i> J.L. LeConte, 1863	DC	MD	-	
Oxyporinae		<i>Oxyporus stygius</i> Say, 1831	DC	MD	VA	
Oxyporinae		<i>Oxyporus vittatus</i> Gravenhorst, 1802	DC	MD	VA	
Oxyporinae		<i>Pseudoxyporus lateralis</i> (Gravenhorst, 1802)	DC	MD	VA	
Oxyporinae		<i>Pseudoxyporus occipitalis</i> (Fauvel, 1864)	DC	MD	VA	
Oxyporinae		<i>Pseudoxyporus quinque maculatus</i> (J.L. LeConte, 1863)	DC	MD	VA	

Oxytelinae	Blediini	<i>Bledius analis</i> J.L. LeConte, 1863	DC	MD	-
Oxytelinae	Blediini	<i>Bledius annularis</i> J.L. LeConte, 1863	DC	-	-
Oxytelinae	Blediini	<i>Bledius assimilis</i> Casey, 1889	-	MD	-
Oxytelinae	Blediini	<i>Bledius basalis</i> J.L. LeConte, 1863	-	MD	-
Oxytelinae	Blediini	<i>Bledius confusus</i> J.L. LeConte, 1877	DC	-	-
Oxytelinae	Blediini	<i>Bledius cordatus</i> (Say, 1831)	-	MD	VA
Oxytelinae	Blediini	<i>Bledius emarginatus</i> (Say, 1831)	DC	MD	VA
Oxytelinae	Blediini	<i>Bledius mandibularis</i> Erichson, 1840	DC	MD	VA
Oxytelinae	Blediini	<i>Bledius neglectus</i> Casey, 1889	-	MD	-
Oxytelinae	Blediini	<i>Bledius nitidicollis</i> J.L. LeConte, 1863	-	MD	-
Oxytelinae	Blediini	<i>Bledius opaculus</i> J.L. LeConte, 1863	-	MD	-
Oxytelinae	Blediini	<i>Bledius philadelphicus</i> Fall, 1919	-	MD	-
Oxytelinae	Blediini	<i>Bledius politus</i> Erichson, 1840	-	MD	-
Oxytelinae	Blediini	<i>Bledius punctatissimus</i> J.L. LeConte, 1877	-	MD	-
Oxytelinae	Blediini	<i>Bledius rubiginosus</i> Erichson, 1840	-	-	VA
Oxytelinae	Blediini	<i>Bledius semiferrugineus</i> J.L. LeConte, 1863	DC	MD	VA
Oxytelinae	Blediini	<i>Bledius sinuatus</i> J.L. LeConte, 1877	DC	-	-
Oxytelinae	Blediini	<i>Bledius tau</i> J.L. LeConte, 1877	-	-	VA
Oxytelinae	Oxytelini	<i>Anotylus densus</i> (Casey, 1893)	-	MD	-
Oxytelinae	Oxytelini	<i>Anotylus exiguus</i> (Erichson, 1840)	DC	-	-
Oxytelinae	Oxytelini	<i>Anotylus insignitus</i> (Gravenhorst, 1806) [†]	DC	-	-
Oxytelinae	Oxytelini	<i>Anotylus nanus</i> (Erichson, 1840)	DC	-	-
Oxytelinae	Oxytelini	<i>Anotylus placusinus</i> (J.L. LeConte, 1877)	DC	-	-
Oxytelinae	Oxytelini	<i>Anotylus rugosus</i> (Fabricius, 1775) [†]	-	-	VA
Oxytelinae	Oxytelini	<i>Anotylus suspectus</i> (Casey, 1893)	DC	-	-
Oxytelinae	Oxytelini	<i>Apocellus sphaericollis</i> (Say, 1831)	DC	-	VA! VA-GWMP
Oxytelinae	Oxytelini	<i>Oxytelus incolumis</i> Erichson, 1840	DC	-	-
Oxytelinae	Oxytelini	<i>Oxytelus invenustus</i> Casey, 1893	-	MD	-
Oxytelinae	Oxytelini	<i>Oxytelus laqueatus</i> (Marsham, 1802) [†]	DC	-	VA
Oxytelinae	Oxytelini	<i>Oxytelus pennsylvanicus</i> Erichson, 1840	DC	-	VA! VA-GWMP
Oxytelinae	Oxytelini	<i>Oxytelus sculptus</i> Gravenhorst, 1806 [†]	DC	-	VA! VA-GWMP

Oxytelinae	Oxytelini			DC	-	VA	
Oxytelinae	Thinobiini	<i>Platystethus americanus</i> Erichson, 1840		DC	-	VA!	VA-GWMP
Oxytelinae	Thinobiini	<i>Carpelinius bilineatus</i> Stephens, 1834†		DC	MD	-	
Oxytelinae	Thinobiini	<i>Carpelinius difficilis</i> (Casey, 1889)		DC	-	-	
Oxytelinae	Thinobiini	<i>Carpelinius fulvipes</i> (Erichson, 1840)		DC	-	-	
Oxytelinae	Thinobiini	<i>Carpelinius gracilis</i> (Mannerheim, 1830)†		DC	-	-	
Oxytelinae	Thinobiini	<i>Carpelinius pusillus</i> (Gravenhorst, 1802)†		DC	-	-	
Oxytelinae	Thinobiini	<i>Carpelinius quadripunctatus</i> (Say, 1831)		DC	MD	VA!	VA-GWMP
Oxytelinae	Thinobiini	<i>Carpelinius rivularis</i> (Motschulsky, 1860)†		-	MD	-	
Oxytelinae	Thinobiini	<i>Carpelinius spretus</i> (Casey, 1889)		DC	MD	-	
Oxytelinae	Thinobiini	<i>Carpelinius subtilis</i> (Erichson, 1839)†		DC	-	-	
Oxytelinae	Thinobiini	<i>Carpelinius uniformis</i> (J.L. LeConte, 1877)		DC	-	-	
Oxytelinae	Thinobiini	<i>Thinobius fimbriatus</i> J.L. LeConte, 1877		DC	-	-	
Oxytelinae	Thinobiini	<i>Thinodromus americanus</i> (Bernhauer & Schubert, 1911)		-	-	VA	
Oxytelinae	Thinobiini	<i>Thinodromus arcifer</i> (J.L. LeConte, 1877)		DC	-	VA!	VA-GWMP
Oxytelinae	Thinobiini	<i>Thinodromus corvinus</i> (Casey, 1889)		DC	-	-	
Paederinae	Lathrobiini	<i>Astenus americanus</i> (Casey, 1905)	Astenina	-	MD	VA	
Paederinae	Lathrobiini	<i>Astenus binotatus</i> (Say, 1823)	Astenina	DC	-	-	
Paederinae	Lathrobiini	<i>Astenus brevipennis</i> (Austin, 1877)	Astenina	-	-	VA	
Paederinae	Lathrobiini	<i>Astenus fusciceps</i> (Casey, 1905)	Astenina	-	-	VA	
Paederinae	Lathrobiini	<i>Astenus longiusculus</i> (Mannerheim, 1830)	Astenina	DC	-	-	
Paederinae	Lathrobiini	<i>Astenus prolixus</i> (Erichson, 1840)	Astenina	DC	-	VA	
Paederinae	Lathrobiini	<i>Echiaster brevicornis</i> (Casey, 1886)	Echiasterina	DC	-	VA	
Paederinae	Lathrobiini	<i>Lathrobium armatum</i> Say, 1830	Lathrobiina	DC	-	-	
Paederinae	Lathrobiini	<i>Lathrobium inornatum</i> (Casey, 1905)	Lathrobiina	DC	-	-	
Paederinae	Lathrobiini	<i>Lathrobium simile</i> J.L. LeConte, 1863	Lathrobiina	DC	-	-	
Paederinae	Lathrobiini	<i>Lathrobium suspectum</i> (Casey, 1905)	Lathrobiina	DC	-	-	
Paederinae	Lathrobiini	<i>Lathrobium virginicum</i> (Casey, 1905)	Lathrobiina	-	-	VA	
Paederinae	Lathrobiini	<i>Lobrathium collare</i> (Erichson, 1840)	Lathrobiina	DC	-	VA!	VA-GWMP
Paederinae	Lathrobiini	<i>Lobrathium emarginatum</i> (Watrous, 1981)	Lathrobiina	-	-	VA	
Paederinae	Lathrobiini	<i>Lobrathium longiusculum</i> (Gravenhorst, 1802)	Lathrobiina	DC	-	VA	
Paederinae	Lathrobiini	<i>Pseudolathra ambigua</i> (J.L. LeConte, 1880)	Lathrobiina	DC	-	VA	

Paederinae	Lathrobiini	Lathrobiina	<i>Pseudolathra americana</i> (Duvivier, 1883)	DC	-	-
Paederinae	Lathrobiini	Lathrobiina	<i>Pseudolathra dimidiata</i> (Say, 1830)	DC	-	-
Paederinae	Lathrobiini	Lathrobiina	<i>Pseudolathra fultarsis</i> (Casey, 1905)	-	-	VA
Paederinae	Lathrobiini	Lathrobiina	<i>Pseudolathra merens</i> (Casey, 1905)	-	-	VA
Paederinae	Lathrobiini	Lathrobiina	<i>Pseudolathra pallidula</i> (J.L. LeConte, 1880)	DC	-	-
Paederinae	Lathrobiini	Lathrobiina	<i>Pseudolathra ventralis</i> (J.L. LeConte, 1880)	DC	-	-
Paederinae	Lathrobiini	Lathrobiina	<i>Tetartopeus angularis</i> (J.L. LeConte, 1863)	DC	-	VA
Paederinae	Lathrobiini	Lathrobiina	<i>Tetartopeus floridanus</i> Casey, 1905	-	-	VA
Paederinae	Lathrobiini	Lathrobiina	<i>Tetartopeus niger</i> (J.L. LeConte, 1863)	-	-	VA
Paederinae	Lathrobiini	Lathrobiina	<i>Tetartopeus nitidulus</i> (J.L. LeConte, 1880)	-	-	VA
Paederinae	Lathrobiini	Lathrobiina	<i>Tetartopeus rubripennis</i> Casey, 1905	DC	-	VA
Paederinae	Lathrobiini	Lathrobiina	<i>Tetartopeus tetricus</i> Casey, 1905	DC	-	-
Paederinae	Lathrobiini	Medonina	<i>Achenomorphus corticinus</i> (Gravenhorst, 1802)	DC	-	VA
Paederinae	Lathrobiini	Medonina	<i>Hypomedon angustus</i> (Casey, 1905)	-	-	VA
Paederinae	Lathrobiini	Medonina	<i>Hypomedon rufipes</i> (Casey, 1905)	-	-	VA
Paederinae	Lathrobiini	Medonina	<i>Lithocharis ochracea</i> (Gravenhorst, 1802) [†]	DC	-	-
Paederinae	Lathrobiini	Medonina	<i>Sciocharis carolinensis</i> Casey, 1905	DC	-	VA! VA-GWMP
Paederinae	Lathrobiini	Medonina	<i>Sciocharis exilis</i> (Erichson, 1840)	-	-	VA! VA-GWMP
Paederinae	Lathrobiini	Medonina	<i>Sunius confluentus</i> (Say, 1831)	DC	-	VA! VA-GWMP, CWH in VMNH
Paederinae	Lathrobiini	Scopaeina	<i>Orus dentiger</i> (J.L. LeConte, 1880)	-	-	VA! VA-CWH in CWH
Paederinae	Lathrobiini	Scopaeina	<i>Scopaeus delicatus</i> Casey, 1905	-	-	VA
Paederinae	Lathrobiini	Scopaeina	<i>Scopaeus exiguus</i> Erichson, 1840	DC	-	-
Paederinae	Lathrobiini	Scopaeina	<i>Scopaeus nitidus</i> (J.L. LeConte, 1863)	DC	-	-
Paederinae	Lathrobiini	Scopaeina	<i>Scopaeus opacus</i> (J.L. LeConte, 1863)	DC	-	-
Paederinae	Lathrobiini	Stilicina	<i>Eustilicus tristis</i> (F.E. Melsheimer, 1844)	DC	-	VA
Paederinae	Lathrobiini	Stilicina	<i>Rugilus angularis</i> (Erichson, 1840)	DC	-	VA! VA-ERH in GWMP+ CWH
Paederinae	Lathrobiini	Stilicina	<i>Rugilus biarmatus</i> (J.L. LeConte, 1880)	DC	-	VA! VA-ERH in VMNH

Paederinae	Lathrobiini	Stilicina	<i>Rugilus ceylanensis</i> (Kraatz, 1859) [†]	-	MD	VA
Paederinae	Lathrobiini	Stilicina	<i>Rugilus dentatus</i> Say, 1831	DC	-	VA! VA-ERH in CWH
Paederinae	Lathrobiini	Stilicina	<i>Rugilus opaculus</i> (J.L. LeConte, 1880)	DC	-	VA
Paederinae	Lathrobiini	Stilicina	<i>Rugilus orbiculatus</i> (Paykull, 1789) [†]	-	-	VA
Paederinae	Lathrobiini	Stilicina	<i>Rugilus rudis</i> (J.L. LeConte, 1863)	DC	-	VA! VA-ERH in VMNH
Paederinae	Lathrobiini	Stilicopsina	<i>Stannoderus monstrosus</i> (J.L. LeConte, 1863)	DC	-	VA
Paederinae	Lathrobiini	Stilicopsina	<i>Stilicopsis paradoxa</i> Sachse, 1852	DC	-	VA! VA-ERH in VMNH
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus badius</i> (Gravenhorst, 1802)	DC	-	VA
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus bicolor</i> (Gravenhorst, 1802)	DC	-	VA
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus capito</i> (Casey, 1885)	-	-	VA
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus carolinus</i> (Erichson, 1840)	DC	MD	VA
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus cinctus</i> (Say, 1830)	DC	-	VA
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus cribratus</i> (J.L. LeConte, 1863)	DC	-	VA! VA-GWMP
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus flavicornis</i> (J.L. LeConte, 1878)	DC	-	-
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus floridanus</i> (J.L. LeConte, 1878)	-	MD	-
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus melanocephalus</i> (Erichson, 1840)	-	-	VA
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus pallipes</i> (Gravenhorst, 1802)	DC	MD	VA
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus parviceps</i> (Casey, 1905)	-	-	VA
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus pimerianus</i> (J.L. LeConte, 1863)	-	-	VA! VA-GWMP
Paederinae	Paederini	Cryptobiina	<i>Homaetarsus virginicus</i> (Casey, 1905)	-	-	VA
Paederinae	Paederini	Cryptobiina	<i>Lissobiops serpentinus</i> (J.L. LeConte, 1863)	DC	-	VA
Paederinae	Paederini	Paederina	<i>Paederus littorarius</i> Gravenhorst, 1806	DC	-	-
Paederinae	Paederini	Paederina	<i>Paederus obliteratus</i> (J.L. LeConte, 1878)	-	MD	VA
Paederinae	Pinophilini	Pinophilina	<i>Lathropinus picipes</i> (Erichson, 1840)	DC	-	VA! VA-CWH in VMNH, AVE in AVE
Paederinae	Pinophilini	Pinophilina	<i>Pinophilus latipes</i> Gravenhorst, 1802	DC	MD	VA
Paederinae	Pinophilini	Pinophilina	<i>Pinophilus opacus</i> J.L. LeConte, 1863	DC	-	VA
Paederinae	Pinophilini	Pinophilina	<i>Pinophilus parvus</i> J.L. LeConte, 1863	-	MD	-
Paederinae	Pinophilini	Proclirina	<i>Palaminus contortus</i> J.L. LeConte, 1878	DC	-	-

Paederinae	Pinophilini	Procirina				
Phloeocharinae			<i>Palaminus testaceus</i> Erichson, 1840	DC	-	-
			<i>Charhyphus picipennis</i> (J.L. LeConte, 1863)	DC	-	VA! VA-CWH in CWH, AVE in AVE
Piestinae			<i>Siagonium americanum</i> (F.E. Melsheimer, 1844)	DC	-	VA! VA-GWMP
Proteininae	Proteinini		<i>Megarthus americanus</i> Sachse, 1852	DC	-	-
Proteininae	Proteinini		<i>Proteinus atomarius</i> Erichson, 1840†	DC	-	-
Pselaphinae	Batrisitae	Amauropini	<i>Arianops jeanneli</i> O. Park, 1956	-	-	VA
Pselaphinae	Batrisitae	Batrisina	<i>Arthmius globicollis</i> J.L. LeConte, 1849	DC	MD	VA
Pselaphinae	Batrisitae	Batrisina	<i>Arthmius involutus</i> Casey, 1893	-	MD!	VA MD-DSC in DSC
Pselaphinae	Batrisitae	Batrisina	<i>Batrisymnoides confinis</i> (J.L. LeConte, 1849)	-	MD	-
Pselaphinae	Batrisitae	Batrisina	<i>Batrisymnoides monstrosus</i> (J.L. LeConte, 1849)	DC	MD	VA
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Babnormodes) antennatus</i> C. Schaeffer, 1906	-	-	VA
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Declivodes) bistriatus</i> (J.L. LeConte, 1849)	DC	MD	-
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Pubimodes) denticollis</i> (Casey, 1884)	DC	MD	VA
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Declivodes) fossicauda</i> (Casey, 1897)	-	-	VA! VA-DSC in CNCI
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Excavodes) frontalis</i> (J.L. LeConte, 1849)	DC!	-	- DC-DSC in USNM
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Excavodes) furcatus</i> (Brendel, 1890)	DC!	MD!	VA! DC-DSC in DEUN; MD-DSC in DSC; VA-GWMP
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Elytroides) ionae</i> (J.L. LeConte, 1849)	DC	MD	VA
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Excavodes) lineaticollis</i> (Aubé, 1833)	DC	MD	VA
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Pubimodes) nigricans</i> (J.L. LeConte, 1849)	DC	-	VA! VA-CWH in VMNH
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Babnormodes) punctifrons</i> (Casey, 1887)	DC	MD	-
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Babnormodes) riparius</i> (Say, 1824)	DC	-	-
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Excavodes) scabriceps</i> (J.L. LeConte, 1849)	DC	MD	VA! VA-GWMP
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Babnormodes) schaefferi</i> O. Park, 1947	-	-	VA
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Spifemodes) schaumii</i> (Aubé, 1844)	DC	MD!	VA MD-DSC in EGRC
Pselaphinae	Batrisitae	Batrisina	<i>Batrisodes (Pubimodes) schmitti</i> (Casey, 1897)	-	-	VA

Pselaphinae	Batritidae	Batritini	Batritina	<i>Batrisodes (Babnormodes) spretus</i> (J.L. LeConte, 1849)	DC	-	VA
Pselaphinae	Batritidae	Batritini	Batritina	<i>Batrisodes (Pubimodes) striatus</i> (J.L. LeConte, 1849)	DC	MD!	VA! MD-DSC in DSC; VA-GWMP
Pselaphinae	Batritidae	Batritini	Batritina	<i>Batrisodes (Excavodes) temporalis</i> (Casey, 1897)	-	MD	-
Pselaphinae	Batritidae	Batritini	Batritina	<i>Batrisodes (Pubimodes) triangulifer</i> (Brendel, 1890)	DC	MD	VA! VA-DSC in DSC
Pselaphinae	Batritidae	Batritini	Batritina	<i>Batrisodes (Babnormodes) unicomis</i> (Casey, 1897)	-	-	VA
Pselaphinae	Batritidae	Batritini	Batritina	<i>Batrisodes (Excavodes) virginiae</i> (Casey, 1884)	-	-	VA
Pselaphinae	Clavigeritae	Clavigerini	Clavigerina	<i>Adranes coecus</i> J.L. LeConte, 1849	-	-	VA
Pselaphinae	Clavigeritae	Clavigerini	Clavigerina	<i>Adranes lecontei</i> Brendel, 1865	DC	-	-
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Euplectus acomanus</i> Casey, 1908	-	MD	-
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Euplectus confluens</i> J.L. LeConte, 1849	DC	MD	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Euplectus disjunctus</i> Casey, 1897	-	-	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Euplectus duryi</i> Casey, 1908	DC	MD	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Euplectus elongatides</i> Newton, 2017	-	MD	-
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Euplectus filiformis</i> (Casey, 1908)	-	-	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Euplectus karstenii</i> (Reichenbach, 1816) [†]	DC	-	-
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Euplectus longicollis</i> Casey, 1884	DC	MD	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Leptoplectus pertenuis</i> (Casey, 1884)	DC	MD	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus congener</i> (Casey, 1884)	DC	-	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus difficilis</i> (J.L. LeConte, 1849)	DC	-	-
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus falcatus</i> J.A. Wagner, 1975	-	-	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus imperfectus</i> Casey, 1897	DC	-	-
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus interruptus</i> (J.L. LeConte, 1849)	DC	-	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus linearis</i> (J.L. LeConte, 1849)	DC	MD!	- MD-DSC in EJFC
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus longipennis</i> Casey, 1908	DC	MD	VA
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus parki</i> J.A. Wagner, 1975	-	MD	-
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus sexualis</i> (Casey, 1884)	-	MD	-
Pselaphinae	Euplectitae	Euplectini	Euplectitae	<i>Pycnoplectus spinifer</i> (Casey, 1884)	-	MD	-
Pselaphinae	Euplectitae	Metopiasini	Rhinoscepsina	<i>Rhinoscepsis (Rhinoscepsis) bistriatus</i> J.L. LeConte, 1878	-	MD!	VA! MD-DSC in DENH; VA-CWH in CWH

Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Biblopectus choctaw</i> Chandler, 1990	-	-	VA!	VA-DSC in DSC
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Biblopectus (Biblopectodes) integer</i> (J.L. LeConte, 1878)	-	-	VA!	VA-DSC in CNCI
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Biblopectus (Biblopectodes) leviceps</i> (Casey, 1884)	DC	MD	VA	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Biblopectus (Biblopectodes) ruficeps</i> (Motschulsky, 1857)	-	-	VA	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Eutyphlus prominens</i> Casey, 1893	-	MD	VA	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Eutyphlus schmitti</i> Raffray, 1904	-	-	VA	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Eutyphlus similis</i> J.L. LeConte, 1880	DC	MD	VA	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Eutyphlus thoracicus</i> O. Park, 1956	-	-	VA	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Pseudactium arcuatum</i> (J.L. LeConte, 1849)	DC	-	-	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Pseudactium parabolicum</i> (Brendel, 1893)	-	-	VA	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Ramecia crinita</i> (Brendel, 1865)	-	-	VA	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Thesiastes fossulatus</i> (Brendel, 1890)	-	MD!	VA!	MD-DSC in CUAC; VA-DSC in DSC
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Thesium cavifrons</i> (J.L. LeConte, 1863)	DC	-	VA!	VA-DSC in EUUC
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Trimioplectus auerbachii</i> O. Park, 1949	DC	-	-	
Pselaphinae	Euplectitae	Trichonychini	Panaphantina	<i>Trimioplectus obsoletus</i> Brendel, 1890	-	-	VA	
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Actiastes foveicollis</i> (J.L. LeConte, 1878)	-	MD	VA	
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Actiastes globifer</i> (J.L. LeConte, 1849)	DC	MD	VA!	VA-DSC in BMNH
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Actiastes wagneri</i> (O. Park, 1963)	-	-	VA	
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Actium impunctatum</i> (Brendel, 1865)	DC	MD	VA	
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Dalmosanus steevesi</i> (Schuster & Grigarick, 1968)	-	MD!	VA	MD-DSC in DSC
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Dalmosella tenuis</i> Casey, 1897	DC	-	VA!	VA-CWH in VMNH
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Melba parvula</i> (J.L. LeConte, 1849)	DC	MD	VA	
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Melba simplex</i> (J.L. LeConte, 1878)	DC	-	-	
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Melba sulcatula</i> Casey, 1897	-	MD	-	
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Melba thoracica</i> (Brendel, 1889)	DC	-	VA	
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Trimiomelba dubia</i> (J.L. LeConte, 1849)	DC	MD	VA	
Pselaphinae	Euplectitae	Trichonychini	Trimina	<i>Trimum brevicorne</i> (Reichenbach, 1816)†	DC	-	-	

Pselaphinae	Euplectidae	Trogastriini	Rhexiina	<i>Rhexius ferrugineus</i> Casey, 1908	DC	MD	VA
Pselaphinae	Euplectidae	Trogastriini	Rhexiina	<i>Rhexius insculptus</i> J.L. LeConte, 1849	DC	-	VA! VA-DSC in MCZC
Pselaphinae	Euplectidae	Trogastriini	Rhexiina	<i>Rhexius schmitti</i> Brendel, 1893	DC	MD	VA
Pselaphinae	Euplectidae	Trogastriini	Rhexiina	<i>Rhexius substriatus</i> J.L. LeConte, 1878	-	MD	VA
Pselaphinae	Euplectidae	Trogastriini	Trogastriina	<i>Conoplectus canaliculatus</i> (J.L. LeConte, 1849)	DC	MD	VA
Pselaphinae	Euplectidae	Trogastriini	Trogastriina	<i>Euborhaxius perscitus</i> (Fletcher, 1932)	-	-	VA
Pselaphinae	Euplectidae	Trogastriini	Trogastriina	<i>Euborhaxius trogasteroides</i> (Brendel, 1892)	-	MD	VA
Pselaphinae	Faronitae			<i>Sonoma holmesii</i> Ferro & Carlton, 2010	-	MD	VA
Pselaphinae	Faronitae			<i>Sonoma streptophorophallus</i> Ferro & Carlton, 2010	-	-	VA
Pselaphinae	Faronitae			<i>Sonoma tischeckini</i> Ferro & Carlton, 2010	-	-	VA! VA-MLF in CWH
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) abdominalis</i> (Aubé, 1833)	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) arguta</i> (Casey, 1897)	DC	MD	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) bellifragi</i> (J.L. LeConte, 1880)	DC	-	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Nisa) cavicornis</i> (Brendel, 1865)	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) corniventris</i> (Motschulsky, 1857)	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) dentata</i> (Say, 1824)	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) floridana</i> (Brendel, 1865)	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) intermedia</i> (Brendel, 1866)	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Nisa) luniger</i> (J.L. LeConte, 1849)	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) shawnee</i> Chandler, 2015	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) terebrata</i> (Casey, 1893)	DC	MD	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) ulkeri</i> (Brendel, 1866)	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Brachygluta (Brachygluta) wickhami</i> Bückle, 2015	DC	-	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Nisaxis tomentosa</i> (Aubé, 1833)	DC	MD	VA! VA-DSC in VMNH
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia atlantica</i> (Brendel, 1866)	DC	-	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia bicolor</i> (Brendel, 1890)	-	MD	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia borealis</i> Casey, 1897	DC!	-	VA DC-DSC in MCZC
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia congener</i> (Brendel, 1865)	DC	-	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia cylindrata</i> (Brendel, 1893)	DC	-	-

Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia demissa</i> Casey, 1893	DC	-	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia divergens</i> (J.L. LeConte, 1880)	DC!	MD	VA DC-DSC in MCZC
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia facilis</i> (Casey, 1884)	-	-	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia gracilis</i> (Casey, 1884)	-	MD	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia hardyi</i> O. Park, 1956	DC	-	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia howardi</i> O. Park, 1958	-	-	VA! VA-DSC in VMNH
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia inopia</i> (Casey, 1884)	DC	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia kansana</i> Casey, 1897	DC	MD!	VA MD-DSC in DSC
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia louisiana</i> Carlton, 2003	-	-	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia peregrinator</i> Casey, 1897	-	MD	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia propinqua</i> (J.L. LeConte, 1849)	-	MD	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia puncticollis</i> (J.L. LeConte, 1849)	DC	MD	VA! VA-DSC in DSC
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia radians</i> (J.L. LeConte, 1880)	DC	MD	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Reichenbachia rubricunda</i> (Aubé, 1844)	DC	-	VA! VA-DSC in MCZC
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Rybaxis clavata</i> (Brendel, 1865)	-	MD	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Rybaxis conjuncta</i> (J.L. LeConte, 1849)	DC	MD	-
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Rybaxis obliquedens</i> Fall, 1927	-	-	VA
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Rybaxis transversa</i> Fall, 1927	-	MD!	- MD-DSC in EJFC
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Rybaxis valida</i> (Brendel, 1890)	DC	-	VA! VA-CWH in VMNH
Pselaphinae	Goniaceritae	Brachyglutini	Brachyglutina	<i>Rybaxis varicornis</i> (Brendel, 1890)	-	-	VA! VA-DSC in DSC
Pselaphinae	Goniaceritae	Brachyglutini	Decarthrina	<i>Decarthron abnorme</i> (J.L. LeConte, 1849)	DC	-	-
Pselaphinae	Goniaceritae	Brachyglutini	Decarthrina	<i>Decarthron exsectum</i> Brendel, 1865	DC	-	-
Pselaphinae	Goniaceritae	Brachyglutini	Decarthrina	<i>Decarthron formiceti</i> (J.L. LeConte, 1849)	DC	-	VA! VA-DSC in DSC
Pselaphinae	Goniaceritae	Brachyglutini	Decarthrina	<i>Decarthron howdeni</i> O. Park, 1956	-	-	VA! VA-DSC in DSC
Pselaphinae	Goniaceritae	Brachyglutini	Decarthrina	<i>Decarthron justum</i> O. Park, 1958	DC	-	VA
Pselaphinae	Goniaceritae	Brachyglutini	Decarthrina	<i>Decarthron longulum</i> (J.L. LeConte, 1849)	DC	-	-
Pselaphinae	Goniaceritae	Brachyglutini	Decarthrina	<i>Decarthron stigmatosum</i> Brendel, 1865	DC	-	VA! VA-DSC in VMNH
Pselaphinae	Goniaceritae	Brachyglutini	Decarthrina	<i>Decarthron velutinum</i> (J.L. LeConte, 1849)	DC	-	-

Pselaphinae	Goniaceritae	Brachyglutini	Eupseniina	<i>Eupsenius dilatatus</i> Motschulsky, 1857	-	-	VA!	VA-DSC in DSC
Pselaphinae	Goniaceritae	Brachyglutini	Pselaptina	<i>Eutrichites zonatus</i> (Brendel, 1865)	DC	MD	VA	
Pselaphinae	Goniaceritae	Bythinini		<i>Machaerodes carinatus</i> (Brendel, 1865)	-	-	VA	
Pselaphinae	Goniaceritae	Bythinini		<i>Tychobythinus carolinae</i> (Casey, 1897)	-	-	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Custotychus carlitaorum</i> Chandler & Geromini, 2016	-	-	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Custotychus daggyi</i> (O. Park, 1949)	DC	MD	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Custotychus lobatus</i> Chandler & Geromini, 2016	-	-	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Custotychus minor</i> (J.L. LeConte, 1849)	DC	-	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Custotychus natchez</i> Chandler & Geromini, 2016	DC	-	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Custotychus pocahontas</i> (Casey, 1897)	DC	MD	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Custotychus verticalis</i> (Casey, 1893)	DC	MD	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Cylindrarctus crinifer</i> Casey, 1893	-	MD!	-	MD-DSC in DSC
Pselaphinae	Goniaceritae	Tychini		<i>Cylindrarctus longipalpis</i> (J.L. LeConte, 1849)	DC	-	VA!	VA-DSC in DSC
Pselaphinae	Goniaceritae	Tychini		<i>Cylindrarctus orientalis</i> Chandler, 1988	DC	-	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Lucifotychus testaceus</i> (Casey, 1884)	-	-	VA	
Pselaphinae	Goniaceritae	Tychini		<i>Nearctitychus sternalis</i> (Raffray, 1904)	-	MD	VA	
Pselaphinae	Pselaphitae	Ctenistini		<i>Atinus monilicornis</i> (Brendel, 1866)	DC	-	VA	
Pselaphinae	Pselaphitae	Ctenistini		<i>Ctenisodes consobrinus</i> (J.L. LeConte, 1849)	DC	-	VA	
Pselaphinae	Pselaphitae	Ctenistini		<i>Ctenisodes lacustris</i> (Casey, 1897)	-	-	VA!	VA-DSC in VMNH
Pselaphinae	Pselaphitae	Ctenistini		<i>Ctenisodes piceus</i> (J.L. LeConte, 1849)	DC	-	-	
Pselaphinae	Pselaphitae	Ctenistini		<i>Ctenisodes zimmermani</i> (J.L. LeConte, 1849)	DC	-	-	
Pselaphinae	Pselaphitae	Pselaphini		<i>Pselaphus bellax</i> Casey, 1893	-	MD	-	
Pselaphinae	Pselaphitae	Pselaphini		<i>Pselaphus erichsoni</i> J.L. LeConte, 1849	DC	-	-	
Pselaphinae	Pselaphitae	Tmesiphorini		<i>Tmesiphorus carinatus</i> (Say, 1824)	DC	MD	VA	
Pselaphinae	Pselaphitae	Tmesiphorini		<i>Tmesiphorus costalis</i> J.L. LeConte, 1849	DC	MD	VA	
Pselaphinae	Pselaphitae	Tyrini	Somatipionina	<i>Hamotus (Hamotoides) opimus</i> Fletcher, 1932	-	-	VA!	VA-DSC in EUUC
Pselaphinae	Pselaphitae	Tyrini	Somatipionina	<i>Upoluna batrisioides</i> (Motschulsky, 1857)	-	-	VA!	VA-CWH in VMNH

Pselaphinae	Pselaphitae	Tyrini	Tyrina	<i>Cedius (Cedius) spinosus</i> J.L. LeConte, 1849	DC	MD	VA!	VA-GWMP, CWH in CWH
Pselaphinae	Pselaphitae	Tyrini	Tyrina	<i>Cedius (Sinistrocedius) ziegleri</i> J.L. LeConte, 1849	DC	MD	VA	
Pselaphinae	Pselaphitae	Tyrini	Tyrina	<i>Ceophyllus monilis</i> J.L. LeConte, 1849	DC	MD	VA	
Pselaphinae	Pselaphitae	Tyrini	Tyrina	<i>Tyrus humeralis</i> (Aubé, 1844)	DC	MD	VA	
Pselaphinae	Pselaphitae	Tyrini	Tyrina	<i>Tyrus semiruber</i> Casey, 1897	-	-	VA!	VA-DSC in VMNH
Pseudopsinae				<i>Pseudopsis subulata</i> Herman, 1975	DC	MD	VA	
Scaphidiinae		Cypariini		<i>Cyparium concolor</i> (Fabricius, 1801)	DC	MD	VA!	VA-GWMP
Scaphidiinae		Scaphidiini		<i>Scaphidium piceum</i> F.E. Melsheimer, 1844	DC	MD	VA	
Scaphidiinae		Scaphidiini		<i>Scaphidium quadriguttatum</i> Say, 1823	DC	MD	VA!	VA-GWMP
Scaphidiinae		Scaphisomatini		<i>Baeocera abdominalis</i> Casey, 1900	-	MD	VA	
Scaphidiinae		Scaphisomatini		<i>Baeocera apicalis</i> J.L. LeConte, 1860	DC	MD	VA	
Scaphidiinae		Scaphisomatini		<i>Baeocera charybda</i> (Cornell, 1967)	-	MD	VA	
Scaphidiinae		Scaphisomatini		<i>Baeocera congener</i> Casey, 1893	DC	-	VA	
Scaphidiinae		Scaphisomatini		<i>Baeocera deflexa</i> Casey, 1893	-	-	VA	
Scaphidiinae		Scaphisomatini		<i>Baeocera falsata</i> Achard, 1920	DC	-	VA	
Scaphidiinae		Scaphisomatini		<i>Baeocera indistincta</i> Löbl & Stephan, 1993	-	MD	-	
Scaphidiinae		Scaphisomatini		<i>Baeocera nana</i> Casey, 1893	DC	MD	VA	
Scaphidiinae		Scaphisomatini		<i>Baeocera picea</i> Casey, 1893	DC	MD	VA	
Scaphidiinae		Scaphisomatini		<i>Baeocera scylla</i> (Cornell, 1967)	-	-	VA	
Scaphidiinae		Scaphisomatini		<i>Baeocera securiforma</i> (Cornell, 1967)	-	-	VA	
Scaphidiinae		Scaphisomatini		<i>Scaphisoma convexum</i> Say, 1825	DC	MD	VA	
Scaphidiinae		Scaphisomatini		<i>Scaphisoma punctulatum</i> J.L. LeConte, 1860	DC	-	-	
Scaphidiinae		Scaphisomatini		<i>Scaphisoma pusillum</i> J.L. LeConte, 1860	DC	-	VA	
Scaphidiinae		Scaphisomatini		<i>Scaphisoma stephani</i> Leschen & Löbl, 1990	-	-	VA	
Scaphidiinae		Scaphisomatini		<i>Scaphisoma suturale</i> J.L. LeConte, 1860	DC	-	-	
Scaphidiinae		Scaphisomatini		<i>Scaphisoma terminatum</i> F.E. Melsheimer, 1844	DC	-	VA!	VA-GWMP
Scaphidiinae		Scaphisomatini		<i>Toxidium gammaroides</i> J.L. LeConte, 1860	DC	MD	VA	
Scydmaeninae	Cephenniitae	Cephenniini		<i>Cephennodes corporosus</i> (J.L. LeConte, 1852)	DC	-	-	
Scydmaeninae	Cephenniitae	Cephenniini		<i>Cephennodes virginicus</i> (Casey, 1897)	-	-	VA	

Scydmaeninae	Cephennitae	Eutheini	<i>Euthiconus latus</i> (Brendel, 1893)	-	MD	VA
Scydmaeninae	Scydmaenitae	Chevrolatiini	<i>Chevrolatia amoena</i> J.L. LeConte, 1866	DC	MD	VA
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Brachycephis subpunctata</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Delius robustulus</i> Casey, 1897	-	-	VA
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) analis</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) basalis</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) brevicornis</i> (Say, 1824)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) calcaratus</i> (Casey, 1897)	-	-	VA
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) capillosulus</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) clavatus</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Euconnus) clavipes</i> (Say, 1824)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Euconnus) consobrinus</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Psomophus) fatuus</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) flavitarsis</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) fossiger</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) fulvus</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) hirtellus</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) humilis</i> (Casey, 1897)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) innocuus</i> (Casey, 1897)	-	-	VA
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Euconnus) lecontei</i> (L.W. Schaufuss, 1866)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) obscurellus</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Euconnus) occultus</i> Casey, 1897	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) pyramidalis</i> (J.L. LeConte, 1863)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Pycnophus) rarus</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Psomophus) salinator</i> (J.L. LeConte, 1852)	DC	MD	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Napochus) trinifer</i> (Casey, 1897)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Euconnus (Euconnus) ventralis</i> Casey, 1897	-	MD	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Lophoderus atlanticus</i> C. Schaeffer, 1905	-	-	VA
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Microscydmus caseyi</i> O'Keefe, 1998	-	MD	VA
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Microscydmus misellus</i> (J.L. LeConte, 1852)	DC	MD	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Neladius tenuis</i> Casey, 1897	-	-	VA

Scydmaeninae	Scydmaenitae	Glandulariini	<i>Stenichnus badius</i> (Casey, 1897)	-	-	VA
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Stenichnus cribrarius</i> (J.L. LeConte, 1852)	DC	-	-
Scydmaeninae	Scydmaenitae	Glandulariini	<i>Stenichnus perforatus</i> (Schaum, 1841)	DC	-	-
Scydmaeninae	Scydmaenitae	Leptoscydmmini	<i>Leptoscydmnus cavifrons</i> Casey, 1897	-	-	VA
Scydmaeninae	Scydmaenitae	Scydmaenini	<i>Scydmaenus grossus</i> (J.L. LeConte, 1863)	DC	-	-
Scydmaeninae	Scydmaenitae	Scydmaenini	<i>Scydmaenus longicollis</i> (Casey, 1897)	-	-	VA
Scydmaeninae	Scydmaenitae	Scydmaenini	<i>Scydmaenus motschulskii</i> (J.L. LeConte, 1863)	DC	-	-
Scydmaeninae	Scydmaenitae	Scydmaenini	<i>Scydmaenus zimmermanni</i> Schaum, 1841	DC	-	-
Staphylininae		Diachini	<i>Diachus schaumii</i> Kraatz, 1860	DC	MD	VA
Staphylininae		Othiini	<i>Atrecus americanus</i> (Casey, 1906)	-	-	VA
Staphylininae		Staphylinini	<i>Acylophorus agilis</i> Smetana, 1971	-	MD	VA!
Staphylininae		Staphylinini	<i>Acylophorus caseyi</i> Leng, 1920	-	-	VA!
Staphylininae		Staphylinini	<i>Acylophorus densus</i> J.L. LeConte, 1878	DC	-	-
Staphylininae		Staphylinini	<i>Acylophorus flavicollis</i> Sachse, 1852	DC	MD	VA
Staphylininae		Staphylinini	<i>Acylophorus lecontei</i> Duvivier, 1883	-	MD	-
Staphylininae		Staphylinini	<i>Acylophorus pronus</i> Erichson, 1840	DC	-	VA!
Staphylininae		Staphylinini	<i>Anaquedius vernix</i> (J.L. LeConte, 1878)	-	MD	VA
Staphylininae		Staphylinini	<i>Hemiquedius infinitus</i> Brunke & Smetana, 2017	DC	-	VA!
Staphylininae		Staphylinini	<i>Heterothops pusio</i> J.L. LeConte, 1863	DC	-	-
Staphylininae		Staphylinini	<i>Tympanophorus puncticollis</i> (Erichson, 1840)	DC	-	VA!
Staphylininae		Staphylinini	<i>Erichsonius alumnus</i> J.H. Frank, 1975	DC	MD	VA
Staphylininae		Staphylinini	<i>Erichsonius brachycephalus</i> J.H. Frank, 1975	-	-	VA
Staphylininae		Staphylinini	<i>Erichsonius civicus</i> J.H. Frank, 1975	-	MD	-
Staphylininae		Staphylinini	<i>Erichsonius nanus</i> (Horn, 1884)	-	-	VA
Staphylininae		Staphylinini	<i>Erichsonius parvus</i> (Horn, 1884)	DC	-	VA
Staphylininae		Staphylinini	<i>Erichsonius patella</i> (Horn, 1884)	DC	-	VA
Staphylininae		Staphylinini	<i>Erichsonius pusio</i> (Horn, 1884)	-	-	VA
Staphylininae		Staphylinini	<i>Erichsonius rosellus</i> J.H. Frank, 1975	-	MD	-
Staphylininae		Staphylinini	<i>Erichsonius rusticus</i> J.H. Frank, 1975	-	MD	-
Staphylininae		Staphylinini	<i>Belonuchus rufipennis</i> (Fabricius, 1801)	DC	MD	VA

Staphylininae	Staphylinini	Philonthina	<i>Bisnius blandus</i> (Gravenhorst, 1806)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Bisnius cephalotes</i> (Gravenhorst, 1802) [†]	DC	MD	-
Staphylininae	Staphylinini	Philonthina	<i>Bisnius inquietus</i> (Erichson, 1840)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Bisnius palmi</i> (Smetana, 1955)	-	MD	-
Staphylininae	Staphylinini	Philonthina	<i>Bisnius pugetensis</i> (Hatch, 1957)	-	-	VA! VA-ERH in GWMP
Staphylininae	Staphylinini	Philonthina	<i>Bisnius siegwaldii</i> (Mannerheim, 1843)	-	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Bisnius sordidus</i> (Gravenhorst, 1802) [†]	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Cafius bistriatus bistriatus</i> (Erichson, 1840)	-	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Gabrius austriacus</i> Scheerpeltz, 1947 [†]	-	MD	-
Staphylininae	Staphylinini	Philonthina	<i>Gabrius fallaciosus</i> (Horn, 1884)	-	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Gabrius microphthalmus</i> (Horn, 1884)	DC	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Gabrius nigrifolius</i> (Gravenhorst, 1802) [†]	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Gabrius ovaliceps</i> (Fall, 1930)	-	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Gabrius splendidulus</i> (Gravenhorst, 1802) [†]	-	MD	-
Staphylininae	Staphylinini	Philonthina	<i>Gabrius vibius</i> Smetana, 1995	-	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Gabronthus thermarum</i> (Aubé, 1850) [†]	DC	-	-
Staphylininae	Staphylinini	Philonthina	<i>Hesperus apicalis</i> (Say, 1830)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Hesperus baltimorensis</i> (Gravenhorst, 1802)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Hesperus stehri</i> Moore, 1958	-	-	VA! VA-GWMP
Staphylininae	Staphylinini	Philonthina	<i>Laetulonthus laetulus</i> (Say, 1830)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Neobisnius jocosus</i> (Horn, 1884)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Neobisnius jucundus</i> (Horn, 1884)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Neobisnius lepidulus</i> (J.L. LeConte, 1863)	DC	-	-
Staphylininae	Staphylinini	Philonthina	<i>Neobisnius occidentoides</i> J.H. Frank, 1981	-	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Neobisnius paederoides</i> (J.L. LeConte, 1863)	DC	MD	VA! VA-GWMP
Staphylininae	Staphylinini	Philonthina	<i>Neobisnius sobrinus</i> (Erichson, 1840)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Neobisnius terminalis</i> (J.L. LeConte, 1863)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus aequalis</i> Horn, 1884	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus asper</i> Horn, 1884	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus caeruleipennis</i> (Mannerheim, 1830)	DC	MD	VA

Staphylininae	Staphylinini	Philonthina	<i>Philonthus carbonarius</i> (Gravenhorst, 1802) [†]	-	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus cautus</i> Erichson, 1840	-	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus cognatus</i> Stephens, 1832 [†]	-	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus concinnus</i> (Gravenhorst, 1802) [†]	-	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus cruentatus</i> (Gmelin, 1790) [†]	-	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus cunctans</i> Horn, 1884	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus debilis</i> (Gravenhorst, 1802) [†]	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus discoideus</i> (Gravenhorst, 1802) [†]	DC	MD	-
Staphylininae	Staphylinini	Philonthina	<i>Philonthus flavibasis</i> Casey, 1915	-	MD	-
Staphylininae	Staphylinini	Philonthina	<i>Philonthus flumineus</i> Casey, 1915	-	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus fusiformis</i> F.E. Melsheimer, 1844	DC	-	-
Staphylininae	Staphylinini	Philonthina	<i>Philonthus gavius</i> Smetana, 1995	-	MD	-
Staphylininae	Staphylinini	Philonthina	<i>Philonthus gracilior</i> Casey, 1915	-	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus hepaticus</i> Erichson, 1840	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus janus</i> Smetana, 1995	-	MD	-
Staphylininae	Staphylinini	Philonthina	<i>Philonthus lornatus</i> Erichson, 1840	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus longicornis</i> Stephens, 1832 [†]	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus monaeses</i> Smetana, 1995	DC	MD	-
Staphylininae	Staphylinini	Philonthina	<i>Philonthus neonatus</i> Smetana, 1965	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus palliatus</i> (Gravenhorst, 1806)	DC	MD	VA! VA-GWMP
Staphylininae	Staphylinini	Philonthina	<i>Philonthus politus</i> (Linnaeus, 1758) [†]	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus quadricollis</i> Horn, 1884	DC	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus rectangularis</i> Sharp, 1874 [†]	-	-	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus rufulus</i> Horn, 1884	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus sericans</i> (Gravenhorst, 1802)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus sericinus</i> Horn, 1884	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus thoracicus</i> (Gravenhorst, 1802)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus umbratilis</i> (Gravenhorst, 1802) [†]	DC	-	-
Staphylininae	Staphylinini	Philonthina	<i>Philonthus umbrinoides</i> Smetana, 1995	-	-	VA! VA-GWMP
Staphylininae	Staphylinini	Philonthina	<i>Philonthus umbrinus</i> (Gravenhorst, 1802)	DC	MD	VA
Staphylininae	Staphylinini	Philonthina	<i>Philonthus validus</i> Casey, 1915	DC	MD	VA

Staphylininae	Staphylinini	Philonthina	<i>Philonthus varians</i> (Paykull, 1789)†	DC	-	-	VA-ERH in GWMP
Staphylininae	Staphylinini	Philonthina	<i>Philonthus ventralis</i> (Gravenhorst, 1802)†	DC	MD	VA!	
Staphylininae	Staphylinini	Quediina	<i>Quedionuchus plagiatus</i> (Mannerheim, 1843)*	-	MD	-	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Distichalius) capucinus</i> (Gravenhorst, 1806)	DC	MD	VA	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Microsaurus) erythrogaster</i> Mannerheim, 1852	-	-	VA	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Microsaurus) fulgidus</i> (Fabricius, 1792)†	DC	MD	VA	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Raphirus) fulvicollis</i> (Stephens, 1833)*	DC	MD	-	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Quedius) laticollis</i> (Gravenhorst, 1802)	DC	MD	VA	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Microsaurus) mesomelinus</i> (Marshall, 1802)†	-	-	VA	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Microsaurus) peregrinus</i> (Gravenhorst, 1806)	DC	MD	VA	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Microsaurus) spelaeus</i> Horn, 1871	-	MD	VA	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Microsaurus) terminatus</i> F.E. Melsheimer, 1844	-	-	VA	
Staphylininae	Staphylinini	Quediina	<i>Quedius (Distichalius) virginicus</i> (Casey, 1915)	-	-	VA	
Staphylininae	Staphylinini	Staphylinina	<i>Creophilus maxillosus villosus</i> (Gravenhorst, 1802)*	DC	MD	VA	
Staphylininae	Staphylinini	Staphylinina	<i>Dinothenarus (Parabenus) badipes</i> (J.L. LeConte, 1863)	-	-	VA	
Staphylininae	Staphylinini	Staphylinina	<i>Ontholestes cingulatus</i> (Gravenhorst, 1802)	DC	MD	VA	
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus caliginosus</i> (Erichson, 1839)	-	-	VA!	VA-AFN in AVE
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus cinnamopterus</i> (Gravenhorst, 1802)	DC	MD	VA	
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus comes</i> (J.L. LeConte, 1863)	DC	MD	VA!	VA-AFN in VMNH
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus exulans</i> (Erichson, 1839)	DC	MD	VA!	VA-AFN in GWMP
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus femoratus</i> (Fabricius, 1801)	-	MD	VA	
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus fossator</i> (Gravenhorst, 1802)	DC	MD	VA!	VA-AFN in VMNH
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus immaculatus</i> (Mannerheim, 1830)	DC	MD	VA!	VA-AFN in USNM
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus maculosus</i> (Gravenhorst, 1802)	DC	MD	VA	
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus mysticus</i> (Erichson, 1840)	DC	MD	VA!	VA-AFN in VPTC
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus pinorum</i> (Casey, 1915)	-	MD	VA!	VA-AFN in USNM
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus praelongus</i> (Mannerheim, 1830)	DC	MD	VA!	VA-AFN in USNM
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus praetermissus</i> Newton, 2011	DC	-	VA	

Staphylininae	Staphylinini	Staphylinina	<i>Platydracus tomentosus</i> (Gravenhorst, 1802)	DC	MD	VA!	VA-AFN in USNM
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus violaceus</i> (Gravenhorst, 1802)	DC	MD	VA!	VA-AFN in GWMP
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus viridanus</i> (Horn, 1879)	DC	MD	VA!	VA-AFN in GWMP
Staphylininae	Staphylinini	Staphylinina	<i>Platydracus zonatus</i> (Gravenhorst, 1802)	DC	MD	VA	
Staphylininae	Staphylinini	Staphylinina	<i>Tasgius (Tasgius) ater</i> (Gravenhorst, 1802) [†]	DC	MD	VA!	VA-AFN in VPTC
Staphylininae	Staphylinini	Staphylinina	<i>Tasgius (Rayachella) winkleri</i> (Bernhauer, 1906) [†]	-	-	VA!	VA-GWMP
Staphylininae	Xantholinini		<i>Gauropterus fulgidus</i> (Fabricius, 1787) [†]	DC	MD	-	
Staphylininae	Xantholinini		<i>Gyrophypnus fracticornis</i> (O.F. Müller, 1776) [†]	DC	MD	VA	
Staphylininae	Xantholinini		<i>Leptacinus pusillus</i> (Stephens, 1833) [†]	DC	-	-	
Staphylininae	Xantholinini		<i>Lithocharodes longicollis</i> (J.L. LeConte, 1863)	DC	MD	VA	
Staphylininae	Xantholinini		<i>Neohypnus attenuatus</i> (Erichson, 1839)	DC	-	-	
Staphylininae	Xantholinini		<i>Neohypnus emmesus</i> (Gravenhorst, 1802)	DC	MD	VA	
Staphylininae	Xantholinini		<i>Neohypnus fragilis</i> (Casey, 1906)	-	-	VA	
Staphylininae	Xantholinini		<i>Neohypnus fusciceps</i> (J.L. LeConte, 1880)	-	-	VA	
Staphylininae	Xantholinini		<i>Neohypnus hamatus</i> (Say, 1830)	DC	-	-	
Staphylininae	Xantholinini		<i>Neohypnus lecontei</i> (Duvivier, 1883)	-	-	VA	
Staphylininae	Xantholinini		<i>Neohypnus melanops</i> (Casey, 1906)	-	MD	VA	
Staphylininae	Xantholinini		<i>Neohypnus obscurus</i> (Erichson, 1839)	-	-	VA	
Staphylininae	Xantholinini		<i>Neohypnus pusillus</i> (Sachse, 1852)	DC	MD	VA	
Staphylininae	Xantholinini		<i>Nudobius cephalus</i> (Say, 1830)	DC	MD	VA	
Staphylininae	Xantholinini		<i>Nudobius luridipennis</i> Casey, 1906	DC	MD	VA	
Staphylininae	Xantholinini		<i>Oxybleptes davisi</i> (Notman, 1924)	DC	-	-	
Staphylininae	Xantholinini		<i>Oxybleptes kiteleyi</i> Smetana, 1982	-	-	VA!	VA-GWMP
Staphylininae	Xantholinini		<i>Phacophallus pallidipennis</i> (Motschulsky, 1858) [†]	-	-	VA	
Staphylininae	Xantholinini		<i>Phacophallus parumpunctatus</i> (Gyllenhal, 1827) [†]	DC	-	-	
Staphylininae	Xantholinini		<i>Stenistoderus rubripennis</i> (J.L. LeConte, 1880)	DC	MD	VA	
Staphylininae	Xantholinini		<i>Xantholinus linearis</i> (A.G. Olivier, 1795) [†]	-	-	VA!	VA-AJB in CWH
Steninae			<i>Stenus alacer</i> Casey, 1884	DC	-	VA	
Steninae			<i>Stenus amabilis</i> (Casey, 1884)	-	MD	VA	

Steninae	<i>Stenus annularis</i> Erichson, 1840	DC	MD	VA
Steninae	<i>Stenus arcullus</i> Erichson, 1840	DC	MD	VA
Steninae	<i>Stenus bilentigatus</i> Casey, 1884	-	-	VA
Steninae	<i>Stenus caenicolus</i> Notman, 1919	-	-	VA
Steninae	<i>Stenus callosifrons</i> Puthz, 1971	-	MD	-
Steninae	<i>Stenus callosus callosus</i> Erichson, 1840	DC	MD	VA
Steninae	<i>Stenus carolinae</i> Casey, 1884	DC	-	-
Steninae	<i>Stenus colon</i> Say, 1831	DC	-	VA
Steninae	<i>Stenus colonus</i> Erichson, 1840	DC	MD	VA
Steninae	<i>Stenus comma</i> J.L. LeConte, 1863*	DC	-	-
Steninae	<i>Stenus compressus</i> Puthz, 1972	-	-	VA
Steninae	<i>Stenus croceatus</i> (Casey, 1884)	DC	-	VA! VA-GWMP
Steninae	<i>Stenus cubensis</i> Bernhauer, 1910	-	MD	VA
Steninae	<i>Stenus delawarensis</i> Casey, 1884	DC	-	-
Steninae	<i>Stenus destitutus</i> Puthz, 2001	-	MD	VA
Steninae	<i>Stenus dispar</i> Casey, 1884	DC	-	-
Steninae	<i>Stenus egenus</i> Erichson, 1840	DC	MD	VA
Steninae	<i>Stenus europs</i> (Casey, 1884)	-	MD	VA
Steninae	<i>Stenus femoratus</i> Say, 1831	DC	-	VA
Steninae	<i>Stenus flavicornis</i> Erichson, 1840	DC	MD	VA
Steninae	<i>Stenus icon</i> Puthz, 1988	-	-	VA
Steninae	<i>Stenus limatulus</i> L. Benick, 1928	-	-	VA
Steninae	<i>Stenus lugens</i> (Casey, 1884)	-	-	VA
Steninae	<i>Stenus lutzi</i> Notman, 1920	DC	MD	VA
Steninae	<i>Stenus meridionalis</i> (Casey, 1884)	-	-	VA
Steninae	<i>Stenus milleporus</i> Casey, 1884	DC	MD	VA
Steninae	<i>Stenus pudicus</i> Casey, 1884	DC	-	VA
Steninae	<i>Stenus punctatus</i> Erichson, 1840	DC	-	VA
Steninae	<i>Stenus reconditus reconditus</i> (Casey, 1884)	-	-	VA
Steninae	<i>Stenus semicolon</i> J.L. LeConte, 1863	DC	-	VA
Steninae	<i>Stenus setiger</i> Puthz, 1971	DC	MD	VA

[illegible]

Tachyporinae	Mycetoporini	<i>Mycetoporus lucidulus</i> J.L. LeConte, 1863	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Cilea silphoides</i> (Linnaeus, 1767) [†]	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Coproporus laevis</i> J.L. LeConte, 1863	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Coproporus ventriculus</i> (Say, 1832)	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Nitidotachinus horni</i> (Campbell, 1973)	-	-	VA!	VA-CWH in CWH
Tachyporinae	Tachyporini	<i>Nitidotachinus scrutator</i> (Gemminger & Harold, 1868)	DC	-	VA!	VA- GWMP, CWH in CWH
Tachyporinae	Tachyporini	<i>Sepedophilus basalis</i> (Erichson, 1839)	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus brachypterus</i> Campbell, 1976	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus campbelli</i> Herman, 2001	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus cinctulus</i> (Erichson, 1839)	-	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus crassus</i> (Gravenhorst, 1802)	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus ctenidialis</i> Campbell, 1976	-	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus debilis</i> (Casey, 1895)	DC	MD	-	
Tachyporinae	Tachyporini	<i>Sepedophilus frosti</i> Campbell, 1976	DC	-	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus kiteleyi</i> Campbell, 1976	-	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus littoreus</i> (Linnaeus, 1758) [†]	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus macer</i> (Casey, 1895)	-	-	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus occultus</i> (Casey, 1885)	DC	MD	VA!	VA-GWMP
Tachyporinae	Tachyporini	<i>Sepedophilus opicus</i> (Say, 1832)	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus parvulus</i> (Horn, 1877)	DC	-	-	
Tachyporinae	Tachyporini	<i>Sepedophilus scriptus</i> (Horn, 1877)	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus testaceus</i> (Fabricius, 1792) [†]	-	MD	-	
Tachyporinae	Tachyporini	<i>Sepedophilus velocipes</i> (Casey, 1895)	-	MD	VA	
Tachyporinae	Tachyporini	<i>Sepedophilus versicolor</i> (Casey, 1885)	DC	MD	VA	
Tachyporinae	Tachyporini	<i>Tachinus addendus</i> Horn, 1877	DC	MD	-	
Tachyporinae	Tachyporini	<i>Tachinus axillaris</i> Erichson, 1839	-	MD	VA	
Tachyporinae	Tachyporini	<i>Tachinus basalis</i> Erichson, 1839*	-	-	VA	
Tachyporinae	Tachyporini	<i>Tachinus canadensis</i> Horn, 1877	-	MD	VA	
Tachyporinae	Tachyporini	<i>Tachinus fimbriatus</i> Gravenhorst, 1802	DC	MD	VA	

Tachyporinae	Tachyporini	<i>Tachinus fumipennis</i> (Say, 1832)	DC	MD	VA
Tachyporinae	Tachyporini	<i>Tachinus limbatus</i> F.E. Melsheimer, 1844	DC	MD	VA
Tachyporinae	Tachyporini	<i>Tachinus luridus</i> Erichson, 1840	DC	MD	VA
Tachyporinae	Tachyporini	<i>Tachinus memnonius</i> Gravenhorst, 1802	DC	MD	VA
Tachyporinae	Tachyporini	<i>Tachinus minimus</i> Campbell, 1973	DC	MD	VA
Tachyporinae	Tachyporini	<i>Tachinus picipes</i> Erichson, 1839	DC	MD	VA
Tachyporinae	Tachyporini	<i>Tachyporus (Tachyporus) elegans</i> Horn, 1877	DC	MD	-
Tachyporinae	Tachyporini	<i>Tachyporus (Tachyporus) jocosus</i> Say, 1832*	DC	-	VA
Tachyporinae	Tachyporini	<i>Tachyporus (Tachyporus) lecontei</i> Campbell, 1979	-	-	VA! VA-GWMP
Tachyporinae	Tachyporini	<i>Tachyporus (Tachyporus) nanus</i> Erichson, 1839	DC	-	VA
Tachyporinae	Tachyporini	<i>Tachyporus (Palporus) nitidulus</i> (Fabricius, 1781)†	-	-	VA
Trichophyinae		<i>Trichophya pilicornis</i> (Gyllenhal, 1810)†	-	-	VA

Probable Cerulean Warbler x Northern Parula Hybrid in Rockbridge County, Virginia in April 2019

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ABSTRACT

We report our sighting of a probable Cerulean Warbler (*Setophaga cerulea*) x Northern Parula (*Setophaga americana*) hybrid that was located in Rockbridge County, Virginia on 20 April and 28 April 2019. Superficially, the bird resembled an after second-year male Cerulean Warbler, but it had several plumage characteristics of an after second-year Northern Parula. Additionally, the hybrid sang a Northern Parula song. This would be the first record of this hybrid in Virginia.

Keywords: Cerulean Warbler, hybrid, Northern Parula, wood warbler.

INTRODUCTION

The wood-warblers (family Parulidae) are a well-known group of small insect-eating birds. Lovette et al. (2010) have presented a recommendation for revising the relationships among the parulids. They used molecular techniques to assess phylogenetic relationships and, of interest to our paper, they show that Cerulean Warblers and Northern Parulas are very closely related. Recently, Trimbath et al. (2019) reported on a genetic analysis of Cerulean Warbler x Northern Parula hybrids from northeastern Ohio. They showed that the two individuals had the same Cerulean Warbler dam and a Northern Parula sire. The presence of hybrids poses interesting questions with respect to speciation and evolution. Hybridization can be due to a number of reasons. Most commonly, there exists a hybrid zone where the ranges of two closely related species who only recently diverged and have retained many similarities interbred. In birds, hybridization has been reported in many families (Cockrum, 1952) and interestingly there is a high rate of hybridization in waterfowl (Ottenburghs et al., 2016). The presence of hybrids in the Parulidae has long been

recognized; hybrids produced between Golden-winged Warblers and Blue-winged Warblers, Brewster's Warbler or Lawrence's Warbler being the most recognized (Short, 1963). A number of other parulid hybrids have been reported, for example: Northern Waterthrush x Blackpoll Warbler (Short & Robbins, 1967); Black-and-white Warbler x Cerulean Warbler (Parkes, 1978); Orange-crowned Warbler x Nashville Warbler (Ralston et al., 2015); Chestnut-sided Warbler x Magnolia Warbler (Burrell et al., 2016); Cerulean Warbler x Black-throated Blue Warbler (Delancey et al., 2019); Cerulean Warbler x Northern Parula (Trimbath et al., 2019). In this paper, we report on a Cerulean Warbler x Northern Parula hybrid that was discovered in Rockbridge County, Virginia in April 2019.

METHODS

Location

Rockbridge County is located in southwestern Virginia and is bordered by the Blue Ridge Mountains on the east and Appalachian Mountains on the west.

Geographically, Rockbridge County is in the Great Valley of Virginia, and it is situated at the southern end of the Shenandoah Valley. The Maury River, the principal waterway within the county, flows into the James River near the southeastern boundary of the county. The location for the hybrid was along the Maury River south of the city of Buena Vista and along River Road, which parallels the Maury River for several miles. The hybrid was found in an area (37°40'28.33"N; 79°25'31.86"W) with Sycamore (*Platanus occidentalis*) trees bordering a small creek and wet area within 100 meters of the Maury River.

Equipment

The hybrid was photographed using a DSLR camera with a telephoto lens. Sound recordings were made using a Telinga Pro parabolic dish, Sennhieser microphone (MKH 20) and a Fostex FR-2 digital field recorder. In addition, sound recordings of a Cerulean Warbler (ML 85132) and Northern Parula (ML 53302) were obtained with permission from the Macaulay Library at the Cornell Lab of Ornithology. Spectrograms from the hybrid, Cerulean Warbler, and Northern Parula songs were generated using Raven Pro 1.4 (Bioacoustics Research Program, The Cornell Laboratory of Ornithology).

RESULTS AND DISCUSSION

On the morning of 20 April 2019, while birding along River Road and the Maury River in southern Rockbridge Co., Virginia, we discovered the hybrid. The bird was first located by its song, which was recognized by one of us (RAR) as a Northern Parula. As a result, we began looking for a Northern Parula. The bird continued to sing and we were able to locate it in trees. Once we saw the bird, we changed our identification to a Cerulean Warbler based on its plumage presentation. We were somewhat confused, however, because we were hearing a Northern Parula but seeing a Cerulean Warbler. In an effort to get better looks at the bird, we played vocalizations for both Cerulean Warbler and Northern Parula from iBird (version 10.06). At the time, we felt that the hybrid tended to respond to the Cerulean Warbler vocalizations better than the Northern Parula. We photographed the bird (Fig. 1) and later when we reviewed our photographs we realized that there were some inconsistencies with the plumage and our Cerulean Warbler identification. As a result, we returned to the location on 28 April and quickly relocated the bird. On that date, we had the assistance of another observer and were able to record the hybrid singing while confirming that it was the hybrid singing. We used Raven Pro to

create spectrograms of the hybrid's vocalizations (Fig. 2). Additionally, we experimented with playing vocalizations of both the Cerulean Warbler and Northern Parula from iBird and noting the responses of the hybrid. It was clear that the hybrid gave a greater response to the Cerulean Warbler call by flying from perch to perch on different trees singing.

Description of hybrid

Overall, the hybrid (Fig. 1) resembled a Cerulean Warbler exhibiting a blue dorsum and head with a blue breast stripe, white belly and throat, weak blue spotting along the flanks, two prominent white wingbars, and white undertail coverts. The hybrid lacked the yellow coloration seen on the throat and breast of a Northern Parula as well as the orange wash seen below the breast band. The upper mandible was black and the lower mandible was gray, which is consistent with a Cerulean Warbler (Northern Parulas have a black upper mandible and a yellow lower mandible). The hybrid had a clear split eye-ring (a Northern Parula characteristic but can be seen in immature Cerulean Warblers), and a completely blue-gray head and neck instead of the sky-blue coloration seen in Cerulean Warblers. The hybrid lacked the white neck band that extends between the nape and cheek (seen in Cerulean Warblers at all ages), which is consistent with a Northern Parula. Also, some of our photographs appear to show a slight green tinge to the dorsum between the wings (Northern Parulas have a distinct greenish triangle between their wings), but no black longitudinal stripes were visible (a Cerulean Warbler characteristic). In addition, the photographs do not show the distinct black, longitudinal stripes seen in adult Cerulean Warblers. Unfortunately, none of the photographs provide a clear view of the back of the bird.

We recorded a total of seven minutes of the hybrid singing. A review of the spectrograms from our recordings were all consistent with the vocalizations of a Northern Parula. A representative spectrogram from the vocalizations recorded from the hybrid is presented in Fig. 2. The hybrid's spectrogram is consistent with the description of the type A Northern Parula song (see Fig. 3) described by Moldenhaur (2012) as containing a preliminary trill ending with a buzzy trill and inverted chevron terminal note. Cerulean Warblers (see Fig. 4) have an introductory set of longer notes followed by a middle section of shorter notes and ending with a final buzz (Buehler et al., 2013) but they lack the inverted chevron of a Northern Parula. We compared the vocalizations that we recorded to spectrograms from Northern Parula (Fig. 3, ML 53302) and Cerulean Warbler (Fig. 4, ML 85132) and found that the vocalizations from the hybrid are consistent with the



Fig. 1. Photographs of the Cerulean Warbler x Northern Parula hybrid taken on 20 April 2019 in Rockbridge Co., Virginia (photographs by R. A. Rowe).

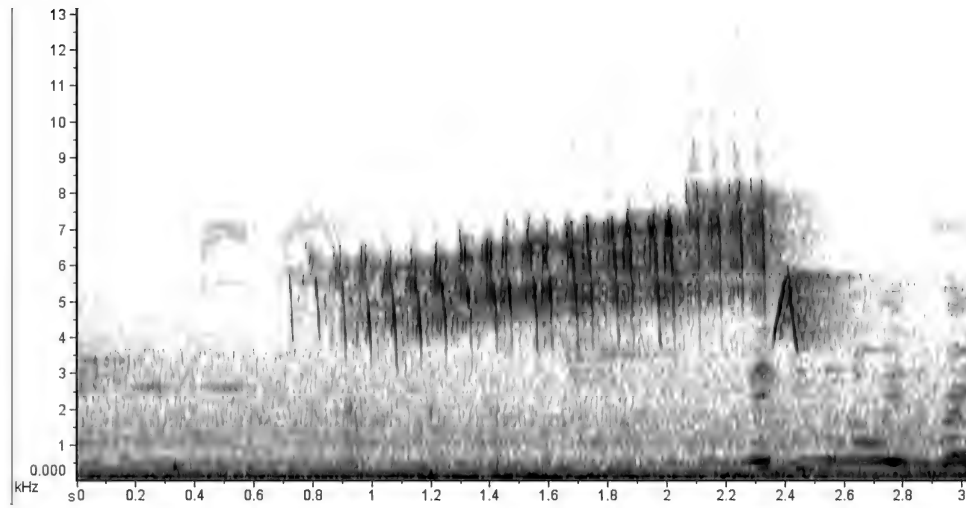


Fig 2. Spectrogram from Cerulean Warbler x Northern Parula hybrid recorded on 28 April 2019 along the Maury River south of Buena Vista, VA. Spectrogram created using Raven Pro 1.4 (Bioacoustics Research Program, Cornell University).

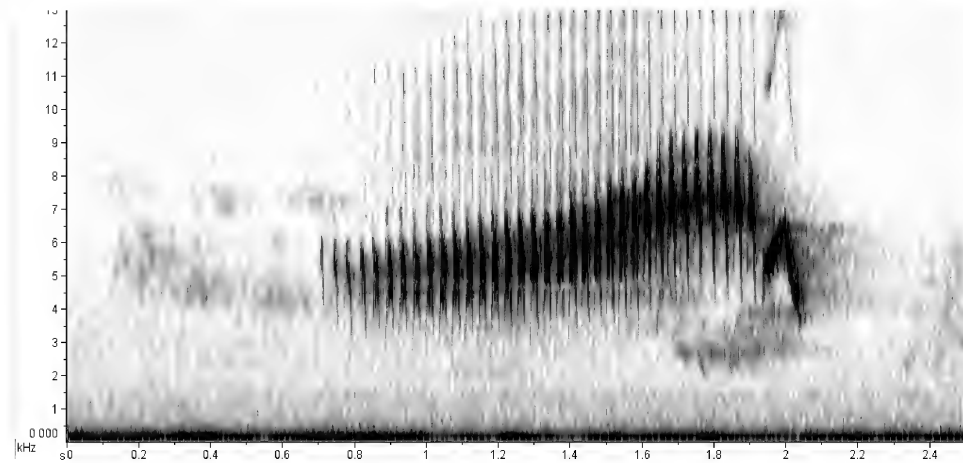


Fig. 3. Spectrogram from a Northern Parula (ML 53302, with permission Macaulay Library at Cornell Lab for Ornithology) created using Raven Pro 1.4 (Bioacoustics Research Program, Cornell University).

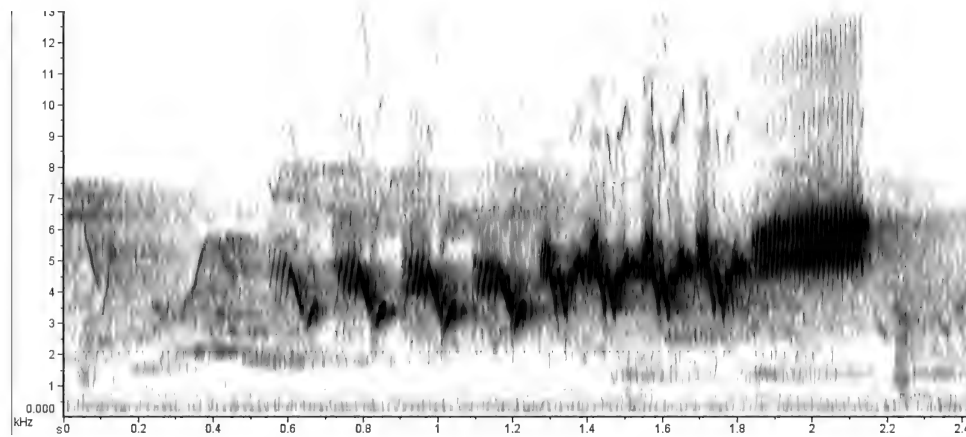


Fig. 4. Spectrogram from a Cerulean Warbler (ML 85132, with permission Macaulay Library at Cornell Lab for Ornithology) created using Raven Pro 1.4 (Bioacoustics Research Program, Cornell University).

vocalizations of a Northern Parula (Fig. 3) and not a Cerulean Warbler (Fig. 4). In addition, we compared our recordings of the hybrid bird to those reported by Trimbath et al. (2019) and note that there is a striking similarity between the two hybrid's vocalizations. If we apply the interpretations of the vocalizations reported by Trimbath et al. (2019) to our recordings, we suspect that our hybrid had a Cerulean Warbler dam and a Northern Parula sire.

There are only a few reports of Cerulean Warbler x Northern Parula hybrids. Pooth & Johnson (2004) reported a suspected hybrid in Dutchess Co., New York, and Nirschl (2004) reported a hybrid in Toledo, Ohio. Lindsey & Vezo (1995) described a suspected interbreeding between a female Cerulean Warbler and a male Northern Parula on Long Island, New York. Trimbath et al. (2019) documented genetic hybridization in two individuals captured in northeastern Ohio. Reports of suspected hybrids have been submitted as photographs and/or sound recordings via eBird to the Macaulay Library at the Cornell Lab of Ornithology. These reports are from various locations with multiple reports from some locations submitted via eBird. In total, there appear to be five individuals being described (Kane, Illinois with photographs or recordings from April and May 2019 at the same location and most likely the same bird: see for example - ML 156727611, 6 May 2019; ML 155845381, 4 May 2019; ML 155532821, 1 May 2019; ML 63231511, 14 May 2017; Cape May, New Jersey ML 139279761, 4 May 2009; ML 63156701, 13 May 2009; Muscatine Iowa ML 59905421, 13 May 2017, Marion Co (Indianapolis), Indiana ML 27567461, 21 April 2016). It is interesting to note that photographs of proposed Cerulean Warbler x Northern Parula hybrids available at the Macaulay Lab show plumage characteristics that are remarkably similar to the bird that we located.

In addition to the Cerulean Warbler x Northern Parula hybrids, other hybrids involving Cerulean Warblers have been reported: Cerulean Warbler x Black-and-white Warbler (Parkes, 1978) and Cerulean Warbler x Black-throated Blue Warbler (Delancey et al., 2019). Hybrids involving Northern Parulas include: Northern Parula x Yellow-throated Warbler, known as Sutton's Warbler (Anich et al., 2012) and Northern Parula x Yellow Warbler (Graves, 1993).

Hybridization in the family Parulidae is not uncommon as shown by the many reports of hybrids in this group. The bird we located exhibited plumage characteristics that are consistent with both Cerulean Warblers and Northern Parulas. Additionally, while the bird superficially resembled a Cerulean Warbler, it sang a Northern Parula song. A few reports of a Cerulean Warbler x Northern Parula hybrid can be found in the

literature or at the Macaulay Library. In these cases, the birds show a mix of plumage characteristics and primarily resemble a Cerulean Warbler. When the song was described or recorded, the hybrids tended to sing a Northern Parula song. Without genetic analysis, we cannot confirm that the bird we discovered was truly a hybrid, but we believe the evidence is compelling that this bird is a Cerulean Warbler x Northern Parula hybrid.

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First Records of the Neotropical Deer Ked *Lipoptena mazamae* Rondani (Diptera: Hippoboscidae) from Virginia

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ABSTRACT

We report the first definitive records of *Lipoptena mazamae* Rondani, a hematophagous ectoparasite of cervids, from the Piedmont of Virginia based on recently collected specimens. The collection localities in Henry and Pittsylvania counties represent the northernmost known occurrence of this species in eastern North America.

Keywords: eastern United States, ectoparasites, range extension, zoonotic diseases.

INTRODUCTION

Louse flies (Diptera: Hippoboscidae) are a small group of obligate hematophagous ectoparasites of birds and mammals represented in the world's fauna by more than 210 species in three subfamilies (Ornithomyinae, Hippoboscinae, and Lipopteninae) (Dick, 2006). These unusual flies are easily recognized by their robust, dorsoventrally flattened bodies, "leathery" appearance, and highly modified antennae and mouthparts (Maa & Peterson, 1987). Most hippoboscids retain functional wings throughout their life, while members of Lipopteninae are either wingless or shed their wings upon locating a host.

Lipopteninae are normally parasitic on cervids (Artiodactyla: Cervidae), while most members of the remaining two subfamilies primarily parasitize birds (Maa, 1969; Maa & Peterson, 1987). Both sexes readily take blood meals and some species are known, or suspected, vectors of etiological agents such as viruses, bacteria, protozoans, and helminths associated with a variety of conditions including bluetongue, West Nile fever, anaplasmosis, bartonellosis, borreliosis, ehrlichiosis, pseudomalaria, and trypanosomiasis (Farajollahi et al., 2005; Small, 2005; de Bruin et al., 2015; Liu et al., 2016; Kelsey & Finch, 2018; Moreira et al., 2019; Skvarla & Machtinger, 2019). The pigeon fly, *Pseudolynchia canariensis* (Macquart), an obligate

parasite of pigeons, is the definitive host and the only known vector of the protozoan *Haemoproteus columbae* Kruse (Moreira et al., 2019). The bluetongue virus is transmitted by the sheep ked, *Melophagus ovinus* (Linnaeus), to its main host the domestic sheep, *Ovis aries* Linnaeus (Small, 2005). In addition, RNA of the West Nile virus was recently detected in *Icosta americana* (Leach) on birds of prey (Farajollahi et al., 2005). Although the actual transfer of pathogens by many of these flies is not completely understood, vertical transmission of *Bartonella* spp. recently has been shown between generations of the sheep ked, *M. ovinus*, and the deer ked, *Lipoptena cervi* (Linnaeus), (Halos et al., 2004; de Bruin et al., 2015).

The subfamily Lipopteninae includes three genera and 34 recognized species (Dick, 2006). Three native (*Lipoptena depressa* [Say], *L. mazamae* Rondani, and *Neolipoptena ferrisi* [Bequaert]) and two introduced (*L. cervi* and *M. ovinus*) species are known from the Nearctic (Maa, 1969; Dick, 2006; Skvarla & Machtinger, 2019). The Neotropical deer ked, *L. mazamae*, the introduced deer ked, *L. cervi*, and the sheep ked, *M. ovinus*, are the only members of Lipopteninae known to occur in eastern North America, the last two of which have been previously reported from Virginia based on verified records (Bequaert, 1957; Small, 2005; Skvarla & Machtinger, 2019). *Lipoptena mazamae* was first reported as occurring in Virginia by Kern (2003),

a record subsequently cited in Skvarla & Machtinger (2019), but neither study included examination of specimens from Virginia (M. J. Skvarla, pers. comm. to LJH, 4 February 2019; W. H. Kern, pers. comm. to LJH, 26 February 2019).

Here we report the first definitive records of *L. mazamae* from Virginia based on recently collected specimens, and discuss the potential range overlap and interaction between *L. mazamae* and *L. cervi*.

MATERIALS

During routine processing of an adult female and a juvenile male White-tailed Deer, *Odocoileus virginianus* (Zimmermann), at the Virginia Museum of Natural History, the first author observed numerous dealate hippoboscids, which were collected, mounted, and subsequently identified as *L. mazamae* (Fig. 1).

The female deer was collected on 1 January 2019, and the entire right rear leg (NDM 4488) was removed and placed in a plastic bag within 30 minutes of the animal's death. The leg was brought to the Virginia

Museum of Natural History and placed in a -20° C freezer on 2 January 2019. On 25 January, the leg was thawed and all of the skin was removed as part of procedures to prepare the bones as an articulated partial skeleton. During skinning, three female deer keds were found concealed in the white fur of the inner leg near the top of the calcaneus. It is unknown whether this was the total number of keds present on the host, as only the right rear leg was available for examination. The host from which these keds were extracted was collected on private property in Pittsylvania Co., Virginia approximately 14 km west of the City of Danville (36.60457°, -79.55312°).

The male deer (NDM 4497) was collected on the morning of 9 October 2019, and the entire carcass was placed in a plastic bag within 12 hours of the animal's death. The carcass was brought to the Virginia Museum of Natural History and placed in a -20° C freezer on the same day. On 19 November the carcass was thawed and the skin was removed as part of procedures to prepare the bones as a skeleton. During skinning, 82 deer keds (39 males, 43 females) were found, most of which were

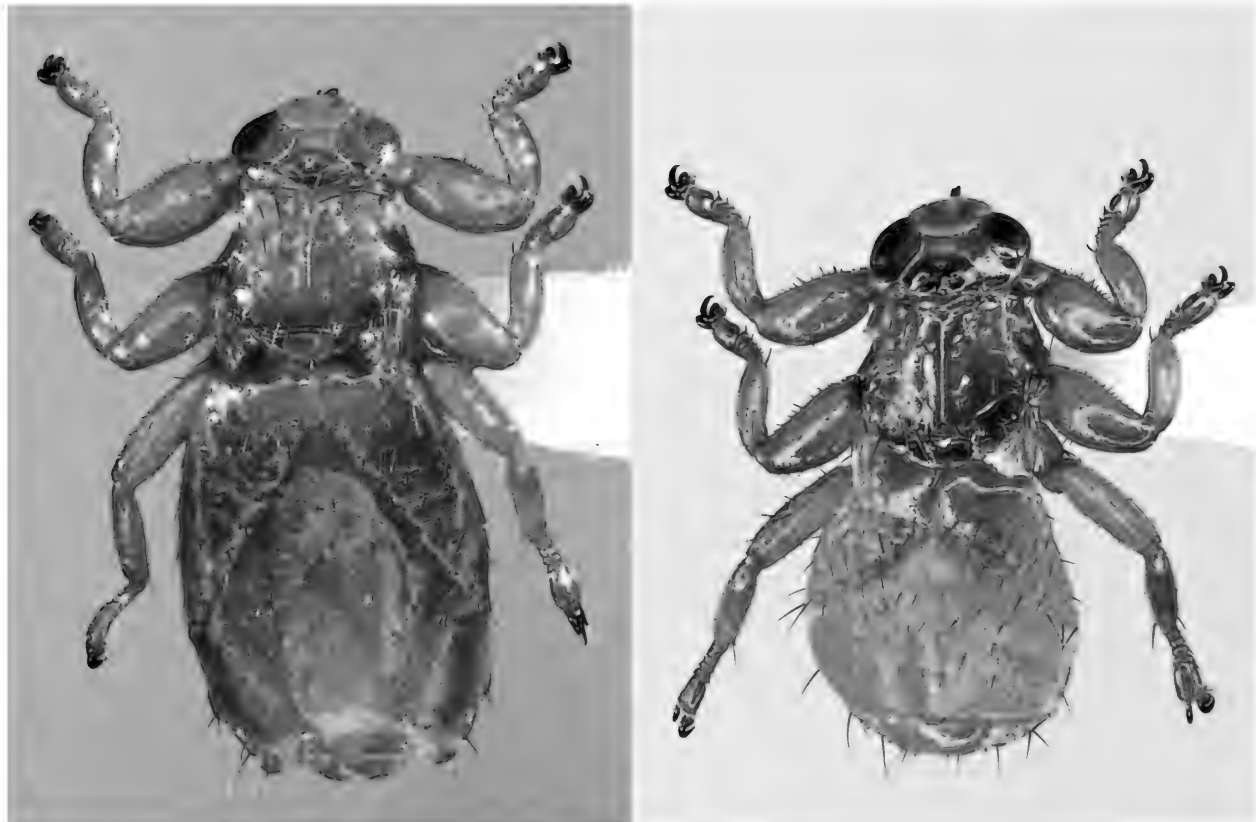


Fig. 1. Dorsal view of (left) dealate female *Lipoptena mazamae* (VMNH 110610) from Pittsylvania Co., Virginia; and (right) dealate male *Lipoptena mazamae* (VMNH 110619.1) from Henry Co., Virginia. Photographs by Lucy Treado, Virginia Museum of Natural History.

concealed in the white fur of the inner rear legs and groin area. Due to poor preservation, only 75 keds (37 males and 38 females) were retained for deposition as specimens. The host from which these keds were extracted was collected on private property in Henry Co., Virginia approximately 8.5 km east of the City of Martinsville (36.67623°, -79.77738°).

All specimens are deposited in the invertebrate collection at the Virginia Museum of Natural History (VMNH 110610-116012 from Pittsylvania Co., and VMNH 110619.1-110619.75 from Henry Co.).

IDENTIFICATION

Members of Lipopteninae are characterized by the fairly dense coverage of evenly spaced spiniform setae on the ventral surface of the thorax; the strongly concave posterior margin of the basal abdominal sternite (convex in *N. ferrisi*); and the greater size of the basal abdominal sternite which is distinctly larger than the scutellum (Maa & Peterson, 1987).

Among the three eastern members of Lipopteninae, *M. ovinus* is easily identified by its rudimentary wings, the absence of halteres, and the reduced eyes, which are each narrower than the antenna. The remaining two eastern taxa are separated based on the position of the median abdominal tergal plates (tergites 4–6 in males, 4–7 in females). In *L. cervi* these are evenly distributed over the abdomen, while they are crowded together in the posterior 1/3 of the abdomen in *L. mazamae* (Fig. 1; see also Figs. 1-5 in Skvarla & Machtinger, 2019).

Keys to the Nearctic Lipopteninae genera and species are found in Maa (1965) and Skvarla & Machtinger (2019).

DISCUSSION

Lipoptena mazamae is an ectoparasite of White-tailed Deer, South American Red Brocket Deer, *Mazama americana* (Erxleben), Central American Red Brocket Deer, *M. temama* (Kerr), and Gray Brocket Deer, *M. gouazoubira* Fisher (Bequaert, 1942; Maa 1969). Incidental hosts include cattle, *Bos taurus* Linnaeus, Tayra, *Eira barbara* (Linnaeus), Peccary, *Pecari tajacu* (Linnaeus), Cougar, *Puma concolor* (Linnaeus), Pampas Deer, *Ozotoceros bezoarticus* (Linnaeus), and humans, *Homo sapiens* Linnaeus (Bequaert, 1942; Bequaert, 1957; Reeves et al., 2006; Graciolli et al., 2011; Shock, 2014). The parasite is commonly found on the legs, abdomen, and genital areas of the host (Samuel & Trainer, 1972). Deer populations may have infestation rates of 60-80%, with individuals sometimes harboring hundreds of keds (Bequaert, 1952; Samuel & Trainer, 1972; Kern, 2003).

The known range of *L. mazamae* includes North, Central, and South America (Kennedy et al., 1987; Skvarla & Machtinger, 2019). South of the United States, this species occurs in Argentina, Bolivia, Brazil, Colombia, Costa Rica, Ecuador, French Guiana, Guatemala, Guyana, Honduras, Mexico, Nicaragua, Panama, Paraguay, Peru, Suriname, Trinidad, Uruguay, and Venezuela (Bequaert, 1957; Maa, 1969; Carreno et al., 2001; Deem et al., 2004; Genoways & Timm, 2005; Romero-Castañón et al., 2008; Graciolli et al., 2011). It reaches as far south as Uruguay and Santiago del Estero in northern Argentina (Bequaert, 1942; Maa, 1969).

In the United States, *L. mazamae* records based on specimens and photo vouchers have been reported from Alabama, Arkansas, Florida, Georgia, Louisiana, Mississippi, Missouri, North Carolina, Oklahoma, South Carolina, Tennessee, and Texas (Skvarla & Machtinger, 2019). The northernmost known localities of *L. mazamae* include St. Clair Co., MO in the West; and Anderson Co., TN and Iredell and Wake counties, NC in the East (Skvarla & Machtinger, 2019).

Our records of *L. mazamae* reported herein may be an indication that this species is naturally expanding its range northward. Climate, and not the absence of its primary host the White-tailed Deer, appears to be the limiting factor for northward expansion of *L. mazamae* (Bequaert, 1954). Bequaert (1954) reported reduction of *L. mazamae* numbers, even to the point of complete absence, during harsh winters when frost was common and below-freezing temperatures were frequent. Given current long-term climate change projections, it is likely that *L. mazamae* will continue to expand its range northward (Collins et al., 2013).

Alternatively, *L. mazamae* may have reached Virginia through human-mediated translocation of its host. White-tailed Deer were nearly extirpated from Virginia in the early 1900s (Handley & Patton, 1947). Between 1930 and 1950, at least 1,800 animals were translocated from Florida, Michigan, North Carolina, Pennsylvania, and Wisconsin into Virginia (Linzey, 1998). Deer from Florida, where the parasite has been present since at least the mid-1930s, could have harbored *L. mazamae* at the time of their relocation (Bequaert, 1935; 1942).

Potential overlap of the geographic ranges of *L. mazamae* and *L. cervi* in Virginia and the surrounding areas was suggested by Skvarla & Machtinger (2019). Our records provide further evidence to support this possibility. Such a scenario represents an interesting ecological situation in which two species that have evolved on different continents might co-occur on the same individual host animal. We can only speculate regarding any potential interactions between the two

taxa, which may range anywhere from exploitation competition to avoidance. It is, however, likely that under such circumstances, the two taxa might exploit different areas of the host's body, a situation previously observed in western deer keds and some ticks (Westrom & Anderson, 1992; Baer-Lehman et al., 2012; Skvarla & Machtinger, 2019).

Actual geographical overlap between the ranges of the two species has not yet been documented. *Lipoptena mazamae* is currently only known from the Piedmont of Virginia while *L. cervi* has been reported from Craig, Montgomery, Shenandoah, and Wythe counties in the Blue Ridge and Ridge & Valley physiographic regions in the mountainous, western part of the state (Skvarla & Machtinger, 2019). Given the close proximity (<80 km) of their currently known ranges, it is possible that a direct physical overlap might occur via natural dispersal of the host and/or the parasites. The annual home ranges of adult male deer in Virginia have been estimated at 2.5 km², with dispersing young moving 6.4 km on average (Batts, 2008). Thus, range expansion of *L. mazamae* and/or *L. cervi* via movements of White-tailed Deer, albeit not rapid, is possible.

Human-mediated dispersal (of host and parasites) represents another potential mechanism for bringing the ranges of the two species together. In most Virginia counties, except for areas in northwestern Virginia where chronic wasting disease has been reported, hunters harvesting deer are allowed to transport entire animals throughout the state (VDGIF, 2019-2020). As deer keds do not leave the host's body for several hours after death of the animal, hunters transporting deer carcasses also are likely to be inadvertently transporting their associated parasites (Samuel & Trainer, 1972).

DNA sequencing of *L. cervi* and its associated hosts has confirmed the presence of five bacterial pathogens (*Anaplasma*, *Bartonella*, *Borrelia*, *Ehrlichia*, and *Rickettsia*) and one protozoan parasite (*Trypanosoma*) in this species (Skvarla & Machtinger, 2019 and references therein). To date, *L. mazamae* has only been tested for and found to contain the Gram-negative bacterium *Bartonella* (Reeves et al., 2006; Souza et al. 2017). Transmission to hosts and spread of pathogens by deer keds is not yet fully understood for any of these disease agents (Skvarla & Machtinger, 2019). However, vertical transmission of *Bartonella schoenbuchensis* corrig. Dehio et al. to offspring has been recently confirmed in *L. cervi*, making it a potential vector for this pathogen (de Bruin et al., 2015).

The close proximity of the currently known ranges of *L. cervi* and *L. mazamae* in Virginia suggests that physical overlap in this state and the surrounding areas is likely, thus setting up an interesting ecological situation. The extent and strength of any potential

interactions between these two species, and the possibility for pathogen exchange between the two taxa, merit further investigation.

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Notes on the Parasitic Beaver Beetle, *Platypsyllus castoris* Ritsema, 1869
and Mouse Nest Beetle, *Leptinus orientamericanus* Peck, 1982
(Coleoptera: Leiodidae: Platypsyllinae) in Virginia

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ABSTRACT

The parasitic Beaver Beetle (*Platypsyllus castoris* Ritsema, 1869) is documented from Virginia for the first time from specimens collected on American Beaver (*Castor canadensis* Kuhl, 1820) in Charles City, Fairfax, and Rappahannock counties. Another leiodid previously recorded from the Commonwealth, the Mouse Nest Beetle (*Leptinus orientamericanus* Peck, 1982), is reported from the Southern Red-backed Vole. *Myodes gapperi* (Vigors, 1830), a new Virginia host record.

Keywords: *Myodes gapperi*, new host record, Southern Red-backed Vole.

INTRODUCTION

Leiodid beetles in the subfamily Platypsyllinae are dorsoventrally flattened and wingless mammalian ectoparasites that lack or have reduced eyes and are associated with rodents, shrews, and moles. The Beaver Beetle, *Platypsyllus castoris* Ritsema, 1869 (Fig. 1), is an ectoparasite of the American Beaver, *Castor canadensis* Kuhl, 1820, in North America and the Eurasian Beaver, *Castor fiber* Linnaeus, 1758, in Eurasia. Originally described as a flea, this species was later recognized as a beetle by LeConte (1872), who placed it in its own family Platypsyllidae, a taxon that was later considered a subfamily of the Leiodidae (Lawrence & Newton, 1982). These eyeless beetles have numerous rows of spines that permit their passage through beaver fur, but help to prevent the host from easily grooming them out.

The American Beaver has a geographic range that extends from Alaska and Canada south of the Arctic Circle to Florida and northern Mexico (Helgen, 2005) and it is likely that the Beaver Beetle has a distribution similar to that of its host (Peck, 2006). The beaver ectoparasite was previously known in North America from seven provinces in Canada (AB, BC, NF, NS, ON, QC, SK) and 26 states in the United States (AK, AL, CA, FL, IA, ID, IL, LA, ME, MI, MN, MO, MS, MT, NC, ND, NE, NJ, NY, OK, OR, PA, SD, TX, WA, WY), the District of Columbia, as well as in Europe (Peck, & Newton, 2017). The specimens noted in this work are the first records of the species' occurrence in Virginia.

MATERIALS AND METHODS

Three beavers killed by vehicles were examined for ectoparasites and their beetles given to Eckerlin who



Fig. 1. *Platypsyllus castoris* Ritsema from Charles City County, Virginia. (credit A.V. Evans)

preserved them in 70% ethanol. Later, they were processed by overnight clearing in 10% potassium hydroxide solution, dehydrated in an ethanol series, cleared in xylene, and mounted to slides in Canada balsam. *Leptinus* beetles were similarly processed and mounted. Vouchers of both species have been deposited in the collections of the National Museum of Natural History (NMNH) and the Virginia Museum of Natural History (VMNH). Specimens collected on the George Washington Memorial Parkway (GWMP) have been deposited in the collections maintained at the GWNP, Turkey Run Park Headquarters, in McLean, Virginia. Three additional Virginia specimens of *Platypsyllus castoris* were collected and identified by Evans and retained in his collection (AVEC) in Richmond, Virginia, which is affiliated with the NMNH in Washington, D.C. All of these beetles were combed out of the fur just behind the shoulders of two beavers. One specimen was point-mounted, while the other two were preserved in 70% ethanol.

RESULTS

The five records of Beaver Beetles collected in Virginia bear the following locality data: Two females ex *Castor canadensis*, 19 January 1986, K.A. Strohmayer, 0.5km south of Belle Haven Marina, GWMP, Fairfax County, Virginia. Five males, two females ex *C. canadensis*, 19 February 1986, J. McBreen, Rt. 123, Lorton, Fairfax County, Virginia. Eight males, three females ex *C. canadensis* March 1986, S.W. Gotte, Lake Royal, Burke, Fairfax County, Virginia. One male ex *C. canadensis*, 4 May 2019, A.V. Evans and M.C. Odom, Harrison Lake National Fish Hatchery, 37.336728° N, 77.187224° W, Charles City County, Virginia. One male and one female ex *C. canadensis*, same collection data as previous record, except 29 August 2019, 37.342805° N, 77.188125° W.

A search for additional Virginia records at the NMNH and VMNH yielded no additional specimens. However, holdings in the Virginia Tech Entomology Collection (VTEC) in Blacksburg, Virginia produced an additional record that included six Beaver Beetles collected in January 1992 by Tom Blount from a beaver in Shenandoah National Park, Rappahannock County, Virginia. These specimens are preserved in ethanol and were identified by Eric Day.

Another ectoparasitic platypsylline, *Leptinus orientamericanus* Peck, 1982, (Fig. 2) occurs in numerous localities in the United States east of the Mississippi River (AL, DC, DE, FL, GA, IL, IN, KY, MD, NC, NJ, NY, OH, PA, SC, TN, VA, WV) and has been recorded in association with seven species of small mammals in the orders Eulipotyphla (formerly Insectivora) and Rodentia (Peck & Newton, 2017). Hosts in Virginia are the Northern Short-tailed Shrew, *Blarina brevicauda* (Say, 1823), Star-nosed Mole, *Condylura cristata* (Linnaeus, 1758), and Eastern Mole, *Scalopus aquaticus* (Linnaeus, 1758) from the counties of Culpeper, Fairfax, Highland, King George, Montgomery, and Tazewell (Peck, 1982; Eckerlin & Painter, 1993).

Eckerlin examined 113 Southern Red-backed Voles, *Myodes gapperi* (Vigors, 1830) in Virginia and found a single female *L. orientamericanus* on a Southern Red-backed Vole collected 2 April 1993, Flattop Mountain, Giles County, Virginia. This specimen, now deposited in the NMNH represents a new Virginia host and county record. Its association with this species is likely accidental simply because of the rarity of its occurrence. Additional specimens of *L. orientamericanus* housed in the VTEC were collected from the Virginia counties of Cumberland, Henry, and Patrick, all of which are new county distribution records.



Fig. 2. *Leptinus orientamericanus* Peck (sex undetermined) from Montgomery County, Indiana. (credit K. Schnepf)

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We thank Steve Gotte, John McBreen, and Karl Strohmayr for providing Beaver Beetle specimens. Michael Odom of the United States Fish and Wildlife Service is acknowledged for his assistance in procuring the specimens of *Platypsyllus castoris* from Charles City County. Kal Ivanov (VMNH), Floyd Shockley (NMNH), Paul Marek and Jackson Means (VTEC) examined their respective collections for additional Virginia records. The Virginia Department of Game and Inland Fisheries kindly provided scientific collecting permits. Kyle Schnepf, Florida State Collection of Arthropods, Gainesville, FL, generously allowed us to use his image of *Leptinus orientamericanus*. Brent Steury offered useful suggestions that contributed to the overall quality of this note.

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Miscellanea

Reports

1. Minutes of the Executive Committee of the Virginia Natural History Society Meeting of November 2, 2019.

The 2019 meeting of the Executive Committee of the Virginia Natural History Society was called to order by President Nancy Moncrief at 9:04 a.m. on November 2, 2019 in the Board Room of the Virginia Museum of Natural History, Martinsville, Virginia. In attendance were Nancy Moncrief, Steve Roble, Kal Ivanov, Karen Powers, Paul Marek, Michael Lachance, Todd Fredericksen, Art Evans, and Curt Harden.

The minutes were approved from the December 1, 2018 Executive Committee Meeting by unanimous consent.

The revisions to Bylaws as edited on December 15, 2018 by Marek, which splits Secretary and Treasurer duties, allows concurrent terms for Councilors, and established Treasurer/co-Treasurer, were approved by unanimous consent.

Announcements:

Transfer of VNHS business office (funds and archive) from Hampden-Sydney to Virginia Museum of Natural History Foundation has been accomplished.

Topics:

Membership Report (Moncrief): As of October 23, there are nine student members, 58 regular members, and nine institutional members for a total of 76 members.

Treasurer Report (Moncrief and Ivanov): On May 10, 2019, VNHS funds (\$17,349.00) were transferred from Hampden-Sydney College to the VNHS business office. The costs of the 2019 Member Meeting were almost covered by the registration fees, and the difference will be paid to VMNH by the VNHS fund. These costs included nametags, poster boards and easels, lunches, and coffee breaks.

Regarding the costs of back issues of *Banisteria*, Ivanov remarked that \$15 per issue is prohibitively costly for students and early-career members. Roble stated that recently-retired faculty might be willing to donate their entire *Banisteria* library to interested parties. Other officers in attendance reminded the others that all

Banisteria issues, excepting the current year, are free online. Ivanov remarked that \$5 per issue (plus cost of shipping) is more reasonable, and if the member wants the entire collection of *Banisteria*, then it would be half off each issue. In addition, the member is obligated to pick up the collection in person from the VMNH archives.

Nomination of officers was not required at this time, and was therefore not discussed.

Date and location of the next joint Executive Committee and Member meetings: Evans mentioned that Randolph-Macon College (in Ashland, near Richmond) is a possibility. Evans argued that by being at a central location would leverage other members in the Commonwealth who might not be compelled to drive to the western part of the state. The Richmond area is good because there are many master naturalists in the area. However, Evans stated that parking is on the street and might be a concern. Randolph-Macon, as a possible location will be researched to determine room rental, lunch choices, street parking, and football scheduling. Moncrief, Powers, and Marek mentioned that the fall works well for their schedules. Powers and Marek stated that continuing the meetings on Saturday is compatible with teaching in the fall. A general discussion ensued about the necessity to avoid a Saturday when a football game at Randolph-Macon would be occurring. Powers mentioned that the Smithsonian center in Front Royal might be a good alternative. Finally, Moncrief mentioned that the 4-H center on Remount Road in Front Royal would be another prospect. (During the discussion with the members in the afternoon member Ellen stated that Charlottesville is yet a fourth possibility of a location for the meeting.) Moncrief remarked that the VMNH is a good location because the museum does not charge VNHS for room rental.

Committee members present identified October 31, 2020 or November 7, 2020 (Saturdays) as appropriate choices of dates. These dates would necessitate a room for 9AM for the next Executive Committee Meeting. Following the Executive Committee meeting, a duration of 10am–4pm would allow 12–14 talks (Ivanov).

Newsletter and Webmaster Report (Marek): Marek requested material for the next newsletter to be sent to him by December 1, 2019. Powers stated that master naturalist coordinator Michelle Prysby would be willing to send the newsletter to master naturalists. Marek will request a list of email addresses of current members from

Moncrief and Ivanov to send the newsletter by January or February 2020. Moncrief stated that the next newsletter in 2020 might contain a section with "President's Remarks". Other items for the 2020 newsletter include a statement that *Banisteria* will be online-only starting in 2020. Moncrief provided revisions to the website to Marek at the Committee Meeting. A new website was necessitated and established for online payment by VNHS. Discussion ensued about transferring the old (.com) website to the new (.org) website with a different URL. Marek expressed concern regarding changing to a new URL since URL permanence in case published bibliographies refer to verbatim *Banisteria* URLs of papers would be broken after one or two years (this would occur even if an auto-redirect was established). Marek will contact Zach at the VMNH about the possibility of porting the payment widget from the new (.org) website to the preexisting (.com) VNHS website.

Editors' Report (Roble and Fredericksen): Roble remarked that he is working on obituaries for *Banisteria* #52. At 9:55AM, the Executive Committee Meeting was tabled until after the Members Conference. At 4:07PM, when the Executive Committee Meeting was reconvened to order, Roble remarked that issue #52 is in the process of being finished, and will be printed and distributed by December 31, 2019. One hundred copies each of issues #52 and 53 will be printed, and they will be mailed together in the same envelope by Roble to all members who have paid dues in 2019 by the date Roble requests the members list from Moncrief and Ivanov. Issue #54 will be entirely online (2020). Marek will inquire with Virginia Tech Libraries regarding DOI generation for the first online issue. Evans recommended that issues 52 and 53 be printed together as a single 52-53 issue. Discussion ensued regarding this solution, and it was resolved that these last print issues will remain separate to demarcate the change of the editor from Roble to Fredericksen. Marek inquired if there would be one or two issues per year when *Banisteria* shifts to entirely online. Fredericksen stated that the published online-only articles each year would receive a DOI and be contained in a separate issue that would be dictated by the year (e.g., all articles published in 2020 will be in issue #54, all articles published in 2021 will be in issue #55, and so on).

Other new business:

Online repository of all the digital files including membership list will be placed on Google Drive.

After the Conference, a Members' meeting was called at 3:38 p.m.

At the members' meeting, Marek solicited feedback from the younger members of the society as to how the VNHS might serve their interests better. One member remarked that a Bioblitz might be held, which are beneficial for communicating to the public the importance of natural history and how scientists collect natural history data. A second member, suggested that bringing more master naturalists into the fold would be beneficial. This member stated that master naturalists like citizen science opportunities, as they can gain volunteer points towards their certification. The member additionally stated that these activities would also be helpful for undergraduates. A third member, recommend that we post the newsletter on the website, and this recommendation was seconded by Evans. A fourth member suggested that we have hands-on activities for the dual purpose of scientific outreach and to provide content for social media. A fifth member, suggested that these hands-on activities might include open labs and natural history collections, and soliciting natural history photos for an online gallery.

The meeting was adjourned at 5:19 p.m.

Respectfully submitted,
Paul E. Marek, Secretary and Webmaster

2. President's Report

The second general meeting of the Virginia Natural History Society (VNHS) was held at the Virginia Museum of Natural History in Martinsville, VA on 2 November 2019. There were 40 attendees, 12 oral presentations, and 10 posters. I am pleased to report that 14 students (including two high school students) presented results of their research projects.

The minutes of the VNHS Executive Committee meeting held on 2 November 2019 are included in this issue. The Committee approved the following changes to the VNHS bylaws: splitting Secretary and Treasurer duties, allowing concurrent terms for Councilors, and establishing Treasurer/co-Treasurer. The revised VNHS Bylaws are also included in this issue.

The Committee also discussed holding the third general meeting of the Society at a location closer to Richmond. Subsequent to the meeting, we learned that 31 October 2020 will probably be the best date for the meeting. We are in communication with a potential venue and will announce the location and confirm the date of the 2020 meeting as soon as possible.

For a variety of financial and logistical reasons, Number 53 will be the last issue of *Banisteria* that will be printed and mailed. Future articles will be published online only. In addition, most or all of the Society's business (announcements, ballots, dues notices) will transition to an electronic format. I encourage members to check the VNHS website frequently for announcements and to contact the VNHS Business Office to confirm that it has their current e-mail address.

Respectfully submitted,
Nancy D. Moncrief, VNHS President

3. Secretary-Treasurer's Report

On 1 May 2019, the VNHS business office was transferred from Hampden-Sydney College (HSC) to the Virginia Museum of Natural History (VMNH), and the VMNH Foundation assumed the role of fiscal agent for VNHS. As of 3 December 2019, total income for 2019 was \$19,714.92, and total expenses were \$2,019.50, leaving an account balance of \$17,695.42.

Details of income are as follows: 1) on 10 May 2019, a total of \$17,349.61 was deposited from the account administered by HSC; 2) dues and contributions from May through 3 December totaled \$1,815.31; 3) registration fees for the 2019 conference totaled \$550.00. Details of expenses are as follows: 1) printing and mailing *Banisteria* 51 totaled \$1,129.70; 2) website fees totaled \$206.93; 3) nametags, easels, and posterboards for conferences totaled \$184.97; 4) lunches and refreshments for the 2019 VNHS conference totaled \$497.90.

Issues 52 and 53 of *Banisteria* are scheduled to go to the printer by 31 December 2019, but those expenses will be paid in 2020.

Respectfully submitted,
Nancy Moncrief and Kal Ivanov, VNHS Co-Treasurers

4. Editor's Report

Banisteria is in a transition phase with a new editor and a new online publication format, starting in 2020 with volume 54. I thank the outgoing editor, Steve Roble, for his many years of service to the Society and his mentorship during the editorial transition.

Online publication format of the journal will save printing costs, increase speed of distribution of the journal to members, and allow for color photographs without costs to authors. Alongside the changes in

publication venue are proposed changes to the editorial structure. First, we would like to extend an invitation to a larger number (10-12) of Associate Editors (nominations are welcomed!), who would be consulted for reviewing or suggesting reviewers within their specialty areas of natural history. Also, a separate section of the journal will be reserved for papers involving citizen-science projects related to natural history. Possible contributors of papers to this section may include Virginia Master Naturalists, high school and college students, and members of nature centers and other non-government institutions. We hope that will increase inclusivity in the Society beyond teaching and research institutions and generate a broader audience to people with an interest in Virginia's natural history. Please contact me if you have any comments or suggestions.

Respectfully submitted,
Todd Fredericksen

Announcements

1. Dues renewal reminder

Please renew your membership for 2020 by paying dues online or by sending a check payable to the Virginia Natural History Society to Co-Treasurer Dr. Nancy Moncrief. The website address for online payments is:

<https://virginiannaturalhistorysociety.com/membership/membership.htm>

You must provide an email address to receive future copies of *Banisteria* and other communications from the society.

Student Member Incentive

The Virginia Natural History Society is offering **free one-year memberships** for students (high school, undergraduate, or graduate) nominated by an advisor/teacher/mentor who is a member in good standing of the Society. This offer is available for up to 20 students each year, and nominations will be considered in the order in which they are received.

Nominators should include the following information for up to three students: name, institution, enrollment level, mailing address, e-mail address, and a short paragraph describing the student's interests in and activities related to Virginia natural history. Nominations should be sent to the Chair of the membership committee, Dr. Kal Ivanov at kal.ivanov@vmnh.virginia.gov.

THE VIRGINIA NATURAL HISTORY SOCIETY
ARTICLES OF INCORPORATION AND BYLAWS

ARTICLES OF INCORPORATION

- Article I. NAME
The society shall be called “The Virginia Natural History Society” (referred to hereinafter as “the Society”).
- Article II. OBJECTIVE
The objective of the Society shall be to promote the study of all aspects of the natural history of Virginia, to educate the citizens of the Commonwealth about natural history, and to conserve its natural resources.
- Article III. The Society shall formulate bylaws to regulate its organization and procedures.
- Article IV. The governing body of the Society shall be the Executive Committee.
- Article V. The Society is organized for scientific, educational, and charitable purposes as defined under the appropriate sections of the Internal Revenue Code. The Executive Committee is empowered to make appropriate changes to retain the tax-exempt status of the Society. Any such changes require a majority vote of the Executive Committee.

BYLAWS

- Article I. MEMBERSHIP
- Section 1. Anyone who is interested in the objective of the Society is eligible for membership. Membership can be attained by application to the Secretary.
- Section 2. The classes of membership shall be Regular, Student, Family, Institutional, Supporting, Patron, Benefactor, Life, and Honorary. Student members must be currently enrolled as graduate, undergraduate, or high school students. A letter from their school verifying their student status must be included with the application for membership. Annual dues for these classes of membership shall be set by the Executive Committee and adjusted as needed. Honorary Councilors pay no dues.
- Section 3. Honorary Councilors shall be nominated by the Executive Committee and voted upon at the Executive Committee Meeting. Additional nominations may be made from the floor. Honorary Councilors shall be elected by a simple majority vote of the Executive Committee. Honorary Councilors shall be recognized leaders in the field of Virginia natural history. No more than 10 Honorary Councilors shall exist at any time. Honorary Councilors shall be eligible to hold office in the Society.
- Section 4. To be a Member in good standing a person must not be in arrears for dues. All annual dues for each ensuing year shall be due 1 January. Publications of the Society will be sent only to Members in good standing and to subscribing Institutions.

Article II. OFFICERS AND COMMITTEES

- Section 1. The officers of the Society shall be a President, a Vice-President (President Elect), a Secretary, a Treasurer, an Editor or co-editors, a webmaster, and three Councilors.
- Section 2. The President shall be the chairman of the Executive Committee and presiding officer at meetings of the Society, and appoint any Standing Committees of the Society and any other committees deemed necessary. The President casts only tie-breaking votes. The President shall serve a 2-year term beginning January 1st and ending December 31st, but will usually not serve for two consecutive terms. The President shall make periodic or annual reports which are published in *Banisteria*.
- Section 3. The Vice-President shall be the President-Elect, shall be elected by the plurality of ballots cast by the Society's membership, and shall succeed to the Presidency in the event the office becomes vacant. The Vice-President shall be chairman of the Membership Committee, shall be responsible for coordinating with the local chairman arrangements for the scientific meetings of the Society, and shall preside over all meetings in the absence of the President.
- Section 4. The Secretary shall be elected by a plurality of the ballots cast by the Society's membership for a 4-year term. The Secretary shall record all the proceedings of the Society, supervise all official mailings including the Society's publications, respond to all inquiries pertaining to membership, subscriptions, and Society matters, and maintain the membership and subscription lists. The Secretary shall compose a summary of the Committee and General Business Meetings.
- Section 4.01 The Treasurer shall be elected by a plurality of the ballots cast by the Society's membership for a 4-year term. The Treasurer shall be in charge of all funds, keep the financial records of the Society, and be responsible for an independent yearly audit. The Treasurer shall summarize the financial health of the Society and submit it for publication in *Banisteria*.
- Section 4.02 The Secretary and Treasurer shall be responsible to the Executive Committee to whom an annual report shall be made.
- Section 5. Editor or Co-editors shall be appointed to a 2-year term of office by the Executive Committee. Editor shall be responsible for preparation of the semiannual journal *Banisteria*, and for maintaining high scholarly standards in its content. The Editor shall be responsible to the Executive Committee to whom an annual report shall be made.
- Section 6. The Webmaster shall maintain and update the Society website and provide an annual report to be published in *Banisteria*.
- Section 7. The Executive Committee shall be the governing body and shall consist of the Executive Council in addition to the President, the Vice-President, the Secretary, the Treasurer, the Editor (or co-editors), Webmaster, and the last three Presidents. The Executive Council shall consist of three Councilors. Three Members in good standing who do not hold other Society offices shall be elected by a plurality of the ballots cast by the Society's membership to serve 4-year terms as councilors. Councilors shall be eligible for reelection 2 years after the expiration of their previous terms.

- Section 8. A quorum of the Executive Committee shall consist of one more than half of its Members and must include the President or Vice-President. Decisions shall be made by simple majority of those Members present.
- Section 9. Vacancies in the staff of officers shall be filled by appointments approved by the majority of the remaining Members of the Executive Committee, except in the case of presidential vacancy (see Article II, Section 3). An appointee shall hold office only for the remainder of the term of his or her predecessor. Interim appointments of officers shall not constitute elected tenure.

Article III. MEETINGS

- Section 1. The Society shall sponsor such Scientific Meetings as it deems advisable. The objectives of the Society's meetings shall be to present appropriate scientific papers and to foster the exchange of ideas among persons interested in Virginia natural history.
- Section 2. There may be an annual General Meeting held sometime during a Scientific Meeting. Notice of this meeting shall be made with the notice of the Scientific Meeting (see Section 6 below). Those present shall constitute a quorum.
- Section 3. The Executive Committee shall meet at some time during each meeting of the Society and at other times if deemed necessary by the President. As occasion demands, the Secretary, at the direction of the President, may submit matters to the Executive Committee for vote by mail or e-mail ballot. All such votes by the Executive Committee shall be placed on record and submitted for ratification at the next meeting of the Executive Committee.
- Section 4. The Vice-President shall recommend the time and place of Annual Scientific Meetings. The Executive Committee shall give final approval to such recommendations.
- Section 5. Notice of a Scientific Meeting shall be published in *Banisteria*, when possible.
- Section 6. All Meetings, except Scientific Meetings, shall be conducted under Robert's Rules of Order.

Article IV. ELECTIONS

- Section 1. The President, Vice-President, Secretary, Treasurer, and Councilors shall be elected by mail or on-line ballot from a list of nominees. Willingness of the nominees to serve shall be secured before their names are placed on the ballot. The Webmaster shall be appointed by the President.
- Section 2. Ballots shall present a choice of at least two candidates for each of the offices of President-Elect, Secretary, Treasurer and Councilor if possible.
- Section 3. The President shall notify the candidates for office of the election results and an announcement of the election results shall be published in the first issue of *Banisteria* following.
- Section 4. A tie vote for any office shall be resolved by a secret ballot of the Executive Committee.

Article V. FINANCES

- Section 1. All funds received by the Society shall be used for publication of all official publications of the Society, and to defray other expenses incurred in the conduct of Society business as determined by the Executive Committee. Society Business includes but is not limited to the following:
- a. Publication of *Banisteria*.
 - b. Publication of meeting programs.
 - c. Rental fees for meeting rooms and monetary support of symposia, bioblitzes etc.
 - d. Registration fees and provision of advanced monies for the purposes of scientific meetings.
 - e. Annual audit.
 - f. Postage and duplicating costs for ballots, reports for the Executive Meeting, and other official mailings of the Society.
 - g. Maintenance of the website and domain.
- Section 2. No part of the net earnings of this Society shall ever inure to, or for the benefit of, or be distributable to its members, trustees, officers, or other private persons, except that the Society shall be empowered to pay reasonable compensation for services rendered, and to make payments and distributions in furtherance of the exempt purposes for which it was formed.
- Section 3. A yearly audit of Society finances shall be made, as provided in Article II, Section 4.
- Section 4. In the event that the Society shall cease to exist, after paying or adequately providing for the debts and obligations of the association, the remaining assets shall be distributed to a non-profit fund, foundation, or corporation, which is organized and operated exclusively for scientific, educational, and/or charitable purposes and which has established its tax exempt status under the appropriate Section of the Internal Revenue Code. The specific non-profit organization or organizations to receive any remaining funds may be determined and approved by the Executive Committee.

Article VI. PUBLICATIONS

The Society shall publish such scientific publications as authorized by the Executive Committee. Members enjoy waived or reduced page charges when publishing in *Banisteria*.

Article VII. DUES

Annual dues shall be determined by the Executive Committee and shall be due before the first day of each year.

Article VIII. FISCAL YEAR

The fiscal year of the Society shall coincide with the calendar year.

Article IX. AMENDMENTS TO THE ARTICLES OF INCORPORATION

- Section 1. Proposed amendments to the Articles of Incorporation may be originated by the Executive Committee or by written request addressed to the Secretary and signed by at least 10 Members or by a simple majority of Members at a General Business Meeting of the Society.
- Section 2. Voting on proposed amendments shall be by mail. The Secretary shall mail copies of proposed amendments and ballots to all members of the Society and shall allow one month for their return, the due date being stated on the ballot. An affirmative vote by two-thirds of the ballots cast by the Society's membership shall be necessary for adoption of an amendment to the Articles of Incorporation. Each proposed amendment shall be accompanied by a concise statement of its purpose, and comparison with the existing provisions, if any.
- Section 3. The Secretary shall count and record the vote on an amendment and shall immediately notify the Executive Committee of the result. The result of the voting shall be announced to the membership of the Society in the next official issue of the journal.

Article X. AMENDMENTS TO THE BYLAWS

Bylaws for the conduct of the business of the Society may be enacted, amended, or repealed by a simple majority vote of the Executive Committee.

Virginia Natural History Society

<http://virginianaturalhistorysociety.com/>

General Information

The Virginia Natural History Society (VNHS) was formed in 1992 to bring together persons interested in the natural history of the Commonwealth of Virginia. The VNHS defines natural history in a broad sense, from the study of plants, animals, and other organisms to the geology and ecology of the state, to the natural history of the native people who inhabit it. The goals of the VNHS are to promote research on the natural history of Virginia, educate the citizens of the Commonwealth on natural history topics, and to encourage the conservation of natural resources.

Dissemination of natural history information occurs through publication of the society's journal *Banisteria* and an annual newsletter. The first issue of *Banisteria*, named in honor of John Banister (1650-1692), the first university-trained naturalist to work in Virginia, was published in 1992. Articles cover a wide array of subjects, and prospective authors are encouraged to submit manuscripts on any aspect of natural history in Virginia; papers may pertain to Virginia or regional archaeology, anthropology, botany, ecology, zoology, paleontology, geology, geography, or climatology. Biographies, obituaries, and historical accounts of relevance to natural history in Virginia are also welcomed. Manuscripts are peer-reviewed for suitability and edited for inclusion in the journal.

The society's website contains detailed instructions for prospective authors and PDF reprints of all *Banisteria* articles that are more than two years old. *Banisteria* is indexed by Zoological Record and is available through EBSCO and the Biodiversity Heritage Library.

Memberships

The VNHS is open to anyone with an interest in natural history and welcomes participation by all members in society activities and efforts to promote education and conservation. Membership includes a subscription to *Banisteria* and invitation to an annual meeting. Annual dues for members are \$20 (per calendar year); library subscriptions are \$40 per year. Payments may be made online or via a check or money order sent to the Treasurer. Copies of most back issues of *Banisteria* are available for sale at a reduced price. The VNHS is a tax-exempt, nonprofit, society under Section 501(C)3 of the IRS. We welcome donations to support our mission in Virginia.

Virginia Natural History Society

Application for Membership

Name _____

Address _____

Zip Code _____

Phone _____

Email _____

Area(s) of Interest _____

ANNUAL DUES AND SUBSCRIPTIONS TO *BANISTERIA*

- ☐ \$500.00 Life (not annual)
- ☐ \$300.00 Benefactor
- ☐ \$100.00 Patron
- ☐ \$50.00 Supporting
- ☐ \$40.00 Institutional
- ☐ \$25.00 Family
- ☐ \$20.00 Regular
- ☐ \$5.00 Student (see below)
- ☐ Contribution in addition to membership dues \$ _____

The special student rate is applicable only when accompanied by the following certification signed by a faculty advisor (**students are also eligible for a 1-year free membership** if an advisor's nomination is approved by the society's Executive Committee; see nomination guidelines in *Banisteria*).

Institution _____

Advisor _____

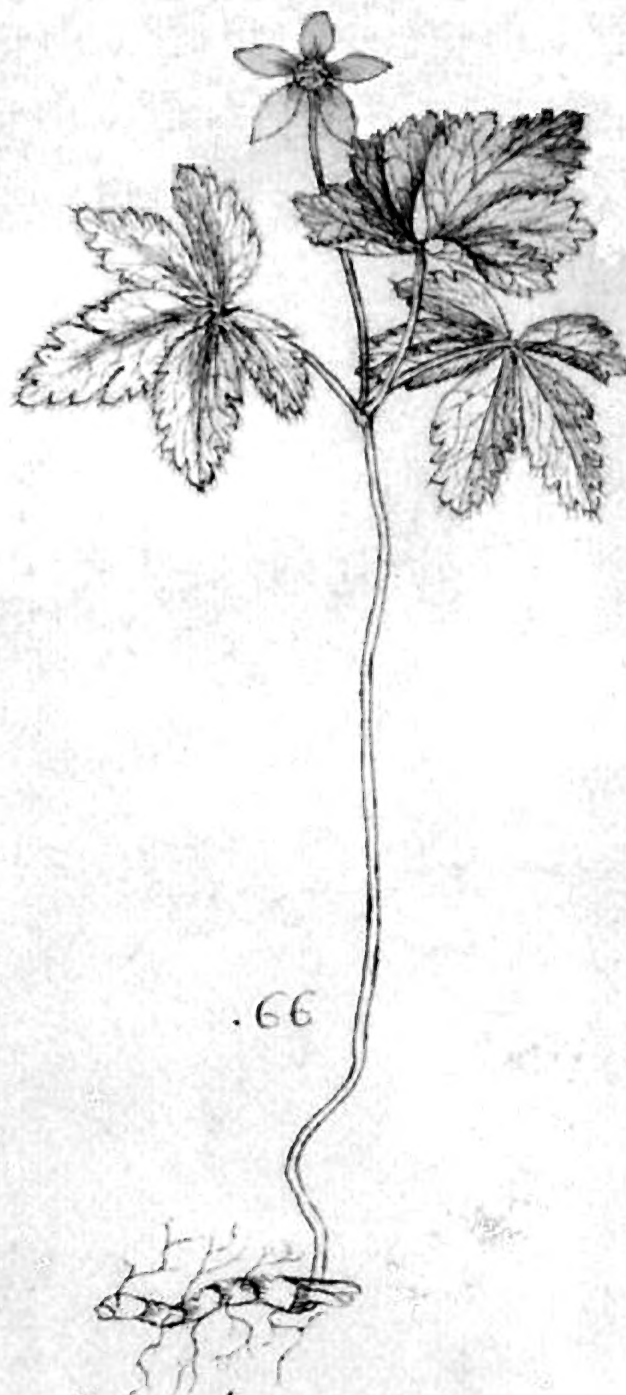
Date _____

Online membership registration and payments may be made at this website:

<https://www.virginianaturalhistorysociety.com/membership/membership.htm>

If paying by mail, send membership form and dues (check or money order payable to Virginia Natural History Society) to:

Dr. Nancy Moncrief, VNHS Treasurer
Virginia Museum of Natural History
21 Starling Avenue
Martinsville, VA 24112



Ranunculus nemorum
Fragaria, folijs.